

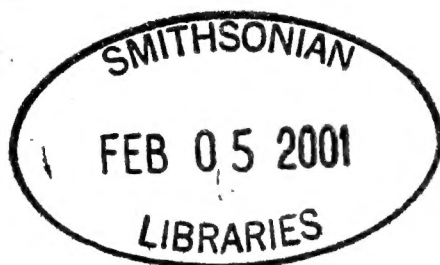
Q
11
K42X
NH

JOURNAL OF THE KENTUCKY ACADEMY OF SCIENCE

Official Publication of the Academy



Volume 61
Number 2
Fall 2000



The Kentucky Academy of Science

Founded 8 May 1914

GOVERNING BOARD EXECUTIVE COMMITTEE 2000

President: Blaine R. Ferrell, Department of Biology, Western Kentucky University, Bowling Green, KY 42101

President Elect: Ron Rosen, Department of Biology, Berea College, Berea, KY 40404

Vice President: Jerry W. Warner, Department of Biological Sciences, Northern Kentucky University, Highland Heights, KY 41099

Past President: Gordon K. Weddle, Department of Biology, Campbellsville University, Campbellsville, KY 42718

Secretary: Stephanie Dew, Department of Biology, Centre College, Danville, KY 40422

Treasurer: William E. Houston, 161 Morningstar Court, Bowling Green, KY 42103

Executive Secretary (ex officio): Donald Frazier, Science Outreach Center, University of Kentucky, Lexington, KY 40536-0078

Editor, JOURNAL (ex officio): John W. Thieret, Department of Biological Sciences, Northern Kentucky University, Highland Heights, KY 41099; (859) 572-6390

Editor, NEWSLETTER (ex officio): Maria K. Falbo, Department of Physics and Geology, Northern Kentucky University, Highland Heights, KY 41099

Chair, Junior Academy of Science (ex officio): Elizabeth K. Sutton, Department of Chemistry, Campbellsville University, Campbellsville, KY 42718

Program Director (ex officio): Robert O. Creek, Department of Biological Sciences, Eastern Kentucky University, Richmond, KY 40475

COMMITTEE ON PUBLICATIONS

Editor and Chair: John W. Thieret, Department of Biological Sciences, Northern Kentucky University, Highland Heights, KY 41099

Associate Editor: James O. Luken, Department of Biological Sciences, Northern Kentucky University, Highland Heights, KY 41099

Index Editor: Varley Wiedeman, Department of Biology, University of Louisville, Louisville, KY 40292

Abstract Editor: Robert Barney, Community Research Service, Kentucky State University, Frankfort, KY 40601

Editorial Board: John P. Harley, Department of Biological Sciences, Eastern Kentucky University, Richmond, KY 40475

Marcus T. McEllistrem, Department of Physics and Astronomy, University of Kentucky, Lexington, KY 40506-0055

J.G. Rodriguez, Department of Entomology, University of Kentucky, Lexington, KY 40546-0091

John D. Sedlacek, Community Research Service, Kentucky State University, Frankfort, KY 40601

Gordon K. Weddle, Department of Biology, Campbellsville University, Campbellsville, KY 42718-2799

All manuscripts and correspondence concerning manuscripts should be addressed to the Editor.

The JOURNAL is indexed in BIOSIS, Cambridge Scientific Abstracts, and in *State Academies of Science Abstracts*.

Membership in the Academy is open to interested persons upon nomination, payment of dues, and election. Application forms for membership may be obtained from the Secretary. The JOURNAL is sent free to all members in good standing.

Annual dues are \$25.00 for Active Members; \$15.00 for Student Members; \$35.00 for Family; \$350.00 for Life Members. Subscription rates for nonmembers are: \$50.00 domestic; \$60.00 foreign. Back issues are \$30.00 per volume.

The JOURNAL is issued semiannually in spring and fall. Two numbers comprise a volume.

Correspondence concerning memberships or subscriptions should be addressed to the Executive Secretary.

⊙ This paper meets the requirements of ANSI/NISO Z39.48-992 (Permanence of Paper).

INSTITUTIONAL AFFILIATES

Fellow

University of Kentucky

University of Louisville

Sustaining Member

Eastern Kentucky University

Northern Kentucky University

Morehead State University

Western Kentucky University

Murray State University

Member

Bellarmino College

Cumberland College

Berea College

Somerset Community College

Campbellsville University

Southeast Community College

Centre College

Associate Member

Georgetown College

Midway College

Jefferson Community College

Owensboro Community College

Kentucky State University

Spalding University

Kentucky Wesleyan College

Thomas More College

Maysville Community College

Transylvania University

INDUSTRIAL AFFILIATES

Associate Patron

Ashland Oil, Inc.

Member

Corhart Refractories Corporation

MPD, Inc.

Associate Member

All-Rite Pest Control

Wood Hudson Cancer Research Laboratory, Inc.

J. Ky. Acad. Sci. 61(2):67-76. 2000.

**Conservation Status and Nesting Biology of the Endangered Duskytail
Darter, *Etheostoma percnurum*, in the Big South Fork of the
Cumberland River, Kentucky**

David J. Eisenhour

Department of Biological and Environmental Sciences, Morehead State University, Morehead, Kentucky 40351

and

Brooks M. Burr

Department of Zoology and Center for Systematic Biology, Southern Illinois University at Carbondale,
Carbondale, Illinois 62901-6501

ABSTRACT

In September 1995 and May and June 1998 and 1999 we conducted an intensive survey of a middle reach of Big South Fork of the Cumberland River in Kentucky with the goal of finding the duskytail darter, *Etheostoma percnurum*, a federally endangered species. Seventy-one specimens were observed in a 19-stream km reach from the mouth of Station Camp Creek, Scott County, Tennessee, to the mouth of Bear Creek, McCreary County, Kentucky. Using underwater observation and a kick-seining technique around slabrocks, we concur with others that the primary habitat of *E. percnurum* includes clear, silt-free pools immediately above riffles where it seeks cover under cobbles and slabrocks. Most Kentucky specimens (31 of 35) and all nests were found in a 3-km reach from just upstream of the mouth of Troublesome Creek to the mouth of Oil Well Branch. On 26 May 1998 five nests were found at two sites. All nests were located immediately above riffles in silt-free glides with slabrock and cobble substrates and were guarded by males. Eggs, deposited in a monolayer on the underside of slabrocks, numbered from 79–103 eggs per nest. Slabrocks with eggs had mean dimensions of $24 \times 19 \times 4.1$ cm and were located in shallow water (51–70 cm) in areas of low flow (5–14 cm/s). Because of the rarity of this darter within its restricted range in Kentucky, we recommend that it be added to Kentucky's list of protected species as endangered. Morphological comparison of *E. percnurum* from across its range revealed that specimens from Big South Fork have more lateral-line scales, are larger, and are shaped differently than specimens from other populations. The morphological and biological comparisons, along with zoogeographic evidence, indicate that the Big South Fork population of *E. percnurum* is an independent evolutionary unit.

INTRODUCTION

Etheostoma percnurum (Perciformes: Percidae), the duskytail darter, is one of 18 species in the darter subgenus *Catonotus* (Braasch and Mayden 1985; Page et al. 1992), a group characterized by a derived spawning habit of clustering eggs in a monolayer on the underside of slabrocks (Braasch and Mayden

1985; Page 1985). It is a member of one of three recognized complexes in the subgenus, the *flabellare* complex, which includes the stripetail darter, *Etheostoma kennicotti*, and the fantail darter, *Etheostoma flabellare*, in addition to *E. percnurum*. The duskytail darter, long known only by its common name, was formally described and distinguished from its

closest relative, the wide-ranging *E. flabellare*, by R. E. Jenkins in 1994 (Jenkins and Burkhead 1994:877–881). At that time, *E. percnurum* was known from six relict populations in drainages of the Cumberland and Tennessee rivers: one in Virginia, Copper Creek; and five in Tennessee, Citico Creek, Abrams Creek, Little River, South Fork Holston River, and Big South Fork of the Cumberland River. Populations in South Fork Holston River and Abrams Creek are believed extirpated (Etnier and Starnes 1993; Jenkins and Burkhead 1994). The only known site of occurrence in the Cumberland River drainage is Big South Fork. Because of this relict distribution, the presumed extirpation of two populations, and threats to water quality in streams it is known to inhabit, *E. percnurum* is listed as Federally Endangered (Biggins 1993). Biggins and Shute (1994), Burkhead and Jenkins (1991), Etnier and Starnes (1993), Jenkins (in Jenkins and Burkhead 1994), Layman (1984, 1991), and Simon and Layman (1995) summarized aspects of life history, development, distribution, and abundance based largely on populations in either Copper Creek or Little River.

Despite numerous and intensive fish collections made over the past 40 years, *E. percnurum* has been reported from only one locality on Big South Fork, the mouth of Station Camp Creek, Scott County, Tennessee. In 1995, we were contracted by the Kentucky Department of Fish and Wildlife Resources (KDFWR) to conduct a status survey of *E. percnurum* in the Kentucky reach of Big South Fork. In this paper we document the status of this species in Kentucky by presenting distributional, abundance, reproduction, and recruitment data. We describe the habitat and nesting biology for the species in Kentucky. Finally, we identify unique morphological, behavioral, and ecological features that indicate the Big South Fork population may be an independent evolutionary unit.

METHODS

Status Survey

From 7–9 Sep 1995 we conducted a nearly comprehensive survey of a middle reach of Big South Fork Cumberland River, Kentucky and Tennessee. We surveyed 14 sites judged to have suitable habitat for *E. percnurum* in a

19-km reach from the mouth of Station Camp Creek, Scott County, Tennessee, to the mouth of Bear Creek, McCreary County, Kentucky. Four of these sites were resampled and two new sites in Big South Fork were surveyed in spring 1998 and 1999 during trips designed to gather information on the nesting biology of *E. percnurum*. Near the Tennessee-Kentucky border, Big South Fork is a medium-sized river, 30–50 m wide, that flows through a deep (200–300 m) gorge of the Cumberland Plateau. Pools are long and deep, with house-sized boulders and bedrock substrates; riffles are fast, well defined, and flow over a substrate of cobbles, boulders, and some pea gravel and coarse sand. The mainstem has a completely forested riparian zone and is protected as a National River and Recreation Area under management of the National Park Service. Because of limited access in this area, nearly all sites required travelling via canoe.

Underwater visual sampling with snorkeling gear was used at all sites for locating individuals of *E. percnurum*. As many as 10 people were involved in underwater sampling at one time, thus increasing the efficiency of the search. In suitable habitat, snorkelers fanned out and turned over slabrocks in pools above and below riffles, macrohabitats known to harbor the species (Jenkins and Burkhead 1994; Layman 1991). This method was supplemented at selected sites by kick-seining (Jenkins and Burkhead 1994) isolated rocks serving as potential cover for *E. percnurum*. About 20–60 minutes of snorkel and/or kick-seining time were spent at each site. Standard physical habitat features (width, depth, area sampled) were recorded at each site. The size of large specimens observed was measured or judged to the nearest millimeter with a plastic ruler. Young-of-the-year (YOY) generally were noted but not measured. Initially, identifications of *E. percnurum* observed by snorkeling were confirmed by capturing individuals with a dipnet. We quickly discovered that even YOY were easily identifiable while we were snorkeling because of the distinctive appearance of *E. percnurum*, the only member of *Catnotus* present in Big South Fork mainstem.

Nesting Biology

Our initial status survey aided in the identification of potential nesting sites for *E. percnurum*.

nurum. Six sites were surveyed for nests in a 27-km reach between Station Camp Creek and Blue Heron, McCreary County, Kentucky, on 25–26 May 1998, 24 Jun 1998, and 18 Jun 1999. Underwater visual observation with snorkeling gear was used to locate nesting adults. Snorkelers concentrated on appropriate habitat above riffles, turning over rocks suitable for use as nesting substrate. About 60–230 minutes of snorkeling time were spent at each site, and up to six people were involved in underwater visual observation. All *E. percunurum* adults observed were captured with dipnets, measured, photographed, and released. The number of eggs in a nest was counted, and the diameter of eggs, nest rocks, and other physical parameters of nest sites were measured with a small plastic ruler or meter tape. Current velocity was measured over the nest with a Swoffer model 2100 flowmeter at 0.6 of the depth above a nest site.

Systematics

To better understand the evolutionary units under protection, we examined and compared specimens from the drainages of the Cumberland and Tennessee rivers. Seventeen meristic and 27 morphometric variables were taken from 65 and 39 specimens, respectively, of *E. percunurum*. Measurements and counts of meristic features follow the methods of Hubbs and Lagler (1974) except that scales above the lateral-line were counted diagonally from the origin of the second dorsal fin. Vertebrae were visualized by the aid of soft x-rays (3A, 30 mv, 15 seconds) and were counted using the methods of Jenkins and Lachner (1971). Cephalic lateral pore counts followed the methods of Page (1983).

Truss-geometric protocol (Humphries et al. 1981; Strauss and Bookstein 1982) was used in part to archive body form and included 17 measurements distributed among three sagittal truss cells with appended anterior and posterior triangles. Ten additional measurements were included in the morphometric analysis. Multivariate analysis of the morphometric data was accomplished using sheared principal component analysis (PCA) (Bookstein et al. 1985; Humphries et al. 1981) to eliminate overall size effects. Principal components were factored from a covariance matrix of log-transformed morphometric variables following the

recommendations of Bookstein et al. (1985). Multivariate analyses were conducted with programs available in SAS 6.01 (SAS Institute, Inc. 1985) and as modified by D. L. Swofford.

Preliminary morphometric analysis revealed strong sexual dimorphism and seasonal variation associated with reproduction in the Copper Creek specimens (the only ones collected in the spring). To reduce confounding variation associated with reproduction, we removed the Copper Creek material from the analysis, and only compared material from collections made outside of the breeding season (August–February).

RESULTS

Status Survey

We observed 60 individuals of *E. percunurum* in September 1995 and 11 individuals in May 1998 in the 19 stream km reach of the Big South Fork between the mouth of Station Camp Creek, Scott County, Tennessee, and the mouth of Bear Creek, McCreary County, Kentucky (Figure 1). Environmental conditions were ideal because the river was at base flow, water clarity was excellent (at least 2 m), direct sunlight was present, and water temperature was warm, averaging 22.2°C in September 1995 and 22.5°C in May 1998. All sites sampled in Tennessee produced 1–10 *E. percunurum*, but only 6 of 10 sites sampled in Kentucky produced individuals, ranging from 1–11 including both adults and YOY (Table 1). Most (31 of 35) individuals observed in Kentucky were in a 3-km reach from just above the mouth of Troublesome Creek to the mouth of Oil Well Branch.

Etheostoma percunurum, not common at any site, was probably the least common darter species observed. Considering that we adequately sampled only 25–50% of the suitable habitat at any one site, the species is presumably more abundant than our results (Table 1) might otherwise suggest. Our professional judgment is that at least 5–10 times the numbers we observed almost certainly inhabit a given site. This conservative estimate would yield a total population of 300–600 individuals of *E. percunurum* in a 19-km stretch of Big South Fork.

Underwater observation proved to be a productive method of finding and observing *E.*

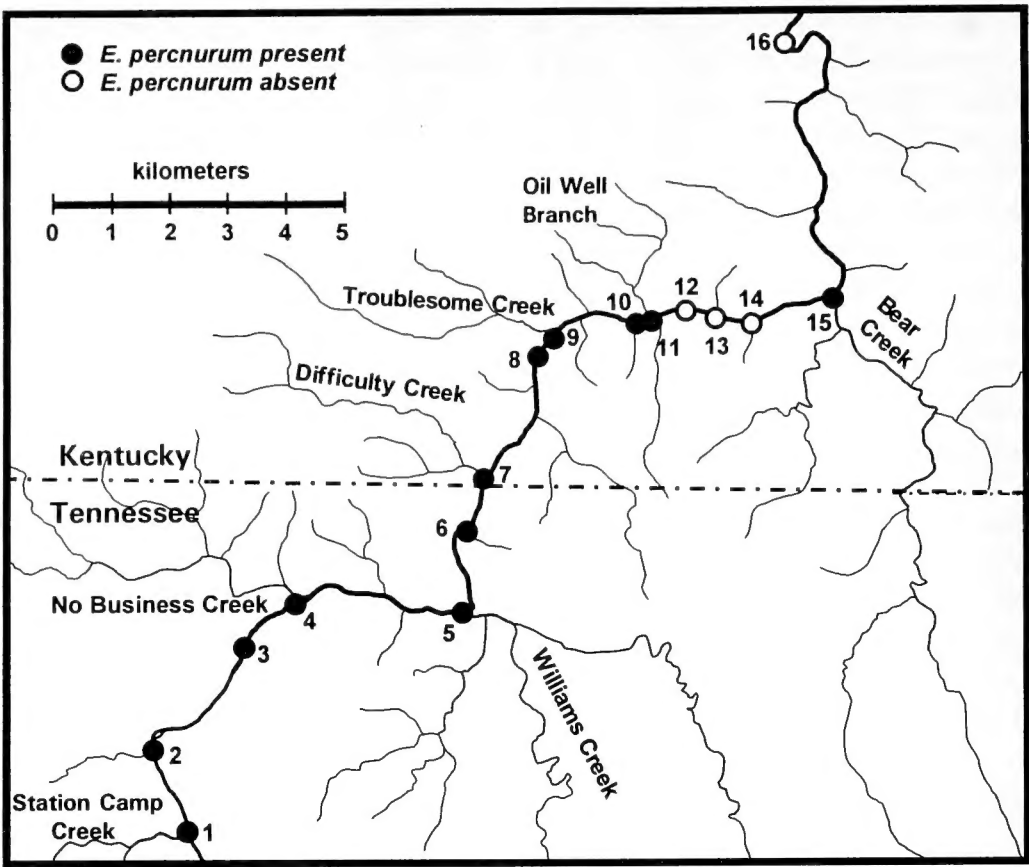


Figure 1. Sampling sites for *Etheostoma percnurum* along Big South Fork of the Cumberland River, Scott County, Tennessee, and McCreary County, Kentucky.

percnurum in most habitats. For comparison, we kick-seined isolated rocks judged to potentially harbor *E. percnurum* at three sites in Kentucky. Initially, our success rate was high, as 3 of 6 rocks sampled yielded *E. percnurum* (site 8). Subsequently, at sites 9 and 11 only 2 of 14 rocks and 1 of 14 rocks sampled, respectively, yielded *E. percnurum*. At these three sites our yield per unit effort for kick-seining (1 fish per 23.3 person-minutes) was considerably higher than for underwater observation (1 fish per 80.8 person-minutes). However, the stacked slabrocks and boulders of most areas precluded sampling by the kick-seining method.

Habitat

All *E. percnurum* were observed in silt-free pools or raceways with low, but evident flow, immediately above riffles where cobbles, boulders, and slabrocks were available. These pools averaged about 25 × 40 m in area and about 54 cm deep, although specimens were observed as deep as 1.5 m. All individuals were under cover of cobbles, boulders, or slabrocks. Cover rocks ranged from fist-sized

cobbles to 76 × 76 cm slabrocks and boulders, with an average thickness of 5 cm. Eleven other darter species were found in association with *E. percnurum*, including *Etheostoma baileyi*, emerald darter; *Etheostoma blennioides*, greenside darter; *Etheostoma camurum*, bluebreast darter; *Etheostoma caeruleum*, rainbow darter; *Etheostoma cinereum*, ashy darter; *Etheostoma sanguifluum*, bloodfin darter; *Etheostoma stigmaeum*, speckled darter; *Etheostoma tippecanoe*, tippecanoe darter; *Etheostoma zonale*, banded darter; *Percina caprodes*, logperch; and *Percina copelandi*, channel darter.

Nesting Biology

Five nests were located in 44.7 person-hours of snorkeling, for a rate of one nest per 8.9 hours of snorkeling. Three nests were found at site 8, just above the mouth of Troublesome Creek, and two nests were found at site 10, mouth of Annie Branch, all on 26 May 1998 (Table 2). All nests were in pools and raceways, 5–50 m above riffles. Nests were in water 51–70 cm (mean = 62 cm) deep with current velocity at 5–14 cm/s (mean = 10 cm/

Table 1. Geographic location, date, number of individuals observed while snorkeling, unit of effort (snorkelers × minutes), and approximate size of specimens of *Etheostoma percnurum*, Big South Fork Cumberland River, Kentucky and Tennessee. Geographic location numbers correspond to those in Figure 1. Sexable adults are divided into males (M) and females (F).

Geographic location	Date	Number of individuals (sex)	Unit of effort	Size of specimens (mm, TL)
1. Mouth of Station Camp Creek, Scott Co., TN	7 Sep 1995	6	9 × 60	35–50 + YOY
	25 May 1998	2F	8 × 30	50
2. Mouth of Parched Corn Creek, Scott Co., TN	7 Sep 1995	6	9 × 60	40–50
3. Halfway between Cold Spring and Big Branch, Scott Co., TN	7 Sep 1995	10	9 × 60	30–50 + YOY
4. Big Island, Scott Co., TN	7 Sep 1995	10	9 × 90	35–50 + YOY
5. Just above mouth of Williams Creek, Scott Co., TN	8 Sep 1995	1	9 × 60	30
6. Near mouth of Hurricane Creek, Scott Co., TN	8 Sep 1995	1	9 × 60	35
7. Mouth of Difficulty Creek, McCreary Co., KY	8 Sep 1995	1	9 × 60	40
	25 May 1998	2	6 × 60	35
8. 1 km above mouth of Troublesome Creek, McCreary Co., KY	8 Sep 1995	11	9 × 110	40–60 YOY
	25–26 May 1998	3M, 1F	6 × 180	50–65
9. Mouth of Troublesome Creek, McCreary Co., KY	9 Sep 1995	7	10 × 75	30–55 + YOY
10. Mouth of Annie Branch, McCreary Co., KY	26 May 1998	2M, 1F	6 × 35	54–67
11. Mouth of Oil Well Branch, McCreary Co., KY	9 Sep 1995	6	10 × 20	40–60 + YOY
12. Huling Ford, McCreary Co., KY	9 Sep 1995	0	10 × 20	—
13. Mouth of second unnamed tributary below Huling Ford, McCreary Co., KY	9 Sep 1995	0	10 × 30	—
14. Mouth of tributary near Slaven’s Branch Trail, McCreary Co., KY	9 Sep 1995	0	10 × 30	—
	26 May 1998	0	2 × 20	—
15. Mouth of Bear Creek, McCreary Co., KY	9 Sep 1995	1	10 × 120	50 mm
16. Blue Heron, McCreary Co., KY	24 Jun 1998	0	2 × 90	—
	18 Jun 1999	0	2 × 150	—

Table 2. Summaries of physical habitat features and nest characteristics of five nests of *Etheostoma percnurum* at sites 8 and 10 (see Figure 1 and Table 1) in Big South Fork Cumberland River, Kentucky, 26 May 1998.

Nest parameter	Mean	Range
Length of nest rock	186 mm	150–300 mm
Width of nest rock	240 mm	180–400 mm
Thickness of nest rock	41 mm	37–50 mm
Depth of nest	62 mm	51–70 mm
Height of nest rock cavity	21 mm	15–25 mm
Size of guardian male	57 mm	55–58 mm
Water temperature	22.5 C	22.5 C
Length and width of egg mass	40 × 50 mm	30 × 40 mm–50 × 75 mm
Diameter of eggs	2–3 mm	2–3 mm
Number of eggs in nest	101	79–132
Current velocity	9.6 cm/s	5.0–14 cm/s

Table 3. Frequency distribution of lateral-line scales in four populations of *Etheostoma percnurum*, Virginia and Tennessee.

Population	39	40	41	42	43	44	45	46	47	48	49	50	n	Mean	SD
Copper Creek, VA	2	1	5	3	5	3	1						20	42.05	1.67
Big South Fork, TN						2	1	1	2	1	—	1	8	46.38	2.07
Little Rock, TN		3	1	5	6	8	4	2	—	1	1		31	43.55	2.08
Citico Creek, TN			1	1	—	1	1	2					6	44.00	2.10

s). All nest rocks were slab-sided and ranged from 15 × 18 cm to 30 × 40 cm and averaged 4.1 cm in thickness. A cavity 15–25 mm deep was between the substrate (sand, coal, and detritus) and the bottom of the nest rock. Nests were located in a relatively small area at each site; nests ranged from 2.4–7.0 m apart. Water temperature was 22.5°C at both sites.

Eggs were adhesive, round, 2–3 mm in diameter, and amber. They were deposited in a monolayer on the underside of the nest rocks except one nest had one egg laid on top of another egg. Four of the five clutches contained “eyed” embryos rapidly moving inside their chorions. The number of eggs in the nest clutches (or the complement of eggs) ranged from 79–132 (mean = 101). Clutches were oblong to round, ranged in size from 30 × 40 mm to 55 × 75 mm, and generally placed near the center of the underside of the nest rock.

Each of the five nests was guarded by a single male with nuptial colors and morphology similar to other members of *Catonotus*. The knobs on the tips of the first dorsal fin were bright gold to amber, and the edges of the pectoral, soft dorsal, and caudal fins were distinctly peppered with black margins. All males had strong vertical bar development on their sides and blackened heads. The bases of their caudal and pectoral fins were light amber to salmon; pelvic fins were an iridescent white. Standard length (SL) for the five guarding males ranged from 55–58 mm (64–67 mm total length). The four adult females (50–54 mm SL) were found under rocks well away from nests. None of the females were swollen with mature ova and apparently already had spawned.

Parasites

In May 1998 we observed black-spot disease in five of six specimens examined closely. The number of black spots ranged from 1–4, except that one female had about 25 spots.

This female was covered in fungus and appeared to be near death. Black-spot disease was present in only three of the eight specimens vouchered in September 1995; the number of black spots ranged from 1–5.

Systematics

The Big South Fork population had higher mean lateral-line scales than other populations (Table 3). Specimens from Citico Creek were distinctive in having fewer principal caudal rays, scales above the lateral line, scales below the lateral line, scales around the caudal peduncle, and lateral-line scales and more pored lateral-line scales (Table 4). Other meristic characters examined showed little intraspecific variation. Principal component analysis of the meristic variables was not informative.

Sheared PCA of the morphometric variables separated individuals from the Cumberland and Tennessee drainages into non-overlapping clusters, with most separation occurring along the sheared PC 2 axis (Figure 2). Examination of loadings indicates Big South Fork specimens have shorter soft dorsal and anal fins, a shorter anal fin base, a more posteriorly placed anal fin, and a more robust body (Table 5). In addition, a larger maximum size was attained by males from Big South Fork (58 mm SL) and Little River (56 mm SL; Etnier and Starnes 1993) than males from Copper Creek (48 mm SL; Jenkins and Burkhead 1994). Females attained a larger maximum size in Big South Fork (maximum 54 mm SL) than Copper Creek (45 mm SL, Jenkins and Burkhead 1994) and Little River (47 mm SL; Layman 1991).

DISCUSSION

Conservation Status

Etheostoma percnurum occupies a greater range in the Big South Fork than previously known. We have established the existence of the species over a 19-stream km reach at six

Table 4. Meristic counts displaying little intraspecific variation of 65 *Etheostoma percnurum* from four populations in the drainages of the Tennessee and Cumberland rivers, Virginia and Tennessee. Means are followed by ranges in parentheses.

Meristic	Copper Creek, VA n = 20	Big South Fork, TN n = 8	Little Rock, TN n = 31	Citico Creek, TN n = 6
Dorsal spines	6.9 (6–8)	7	6.61 (6–7)	6.50 (6–7)
Dorsal rays	11.50 (11–13)	11.75 (11–12)	11.62 (11–13)	11.67 (11–12)
Pectoral rays	12.75 (12–13)	13	12.52 (12–13)	12.67 (12–14)
Pelvic rays	6	6	6	6
Anal spines	1.95 (1–2)	2	2	2
Anal rays	7.25 (6–8)	7.25 (7–8)	7.19 (7–8)	7.50 (7–8)
Principal caudal rays	17.85 (17–18)	17.25 (16–18)	17.87 (17–18)	16.33 (16–17)
Pored lateral-line scales	24.20 (17–28)	27.63 (25–30)	25.94 (23–30)	31.00 (29–34)
Scales above lateral line	6.80 (6–8)	6.63 (6–7)	6.48 (6–7)	6.00 (5–7)
Scales below lateral line	8.95 (7–10)	8.25 (7–9)	8.71 (8–10)	8.00 (7–9)
Scales around caudal peduncle	24.05 (22–27)	23.13 (22–25)	23.52 (21–27)	21.83 (20–25)
Interorbital pores	6.05 (6–7)	6	6	6
Preoperculomandibular pores	10.00 (9–11)	10.13 (10–11)	10.03 (9–12)	9.50 (9–10)
Total vertebrae	33.55 (33–35; n = 11)	33.63 (33–35)	—	—
Precaudal vertebrae	13.82 (13–15; n = 11)	14.13 (13–15)	—	—
Caudal vertebrae	19.73 (19–20; n = 11)	19.50 (18–20)	—	—

sites in the Tennessee reach of Big South Fork (five of these not previously reported) and report it here for the first time from six sites in the Kentucky reach of Big South Fork. A recent survey by Shute et al. (unpublished data) located *E. percnurum* as far upstream as the mouth of Blevins Branch, Tennessee, expanding the known range in the Big South Fork to a 22-stream km reach. However, most Kentucky specimens are known only from a 3-km reach, and additional populations in Kentucky are unlikely to be present. The Big South Fork harbors the only known population of *E. percnurum* in the Cumberland River drainage.

The few other streams in this drainage that might harbor a relict population of this species have been well sampled (Burr and Warren 1986; Etnier and Starnes 1993). Our population estimate (300–600) over a 19-km stretch, though conservative, indicates considerably lower density in Big South Fork than Little River, Tennessee, where Layman (1991) estimated a population of 1023 *E. percnurum* in a 200-m reach in 1983. The highly restricted and localized distribution (mostly in about 3 stream km) of *E. percnurum* in Kentucky as well as its general rarity argue strongly for its immediate inclusion on the Kentucky state endangered/threatened species list as an endangered species.

The small distribution and population size of *E. percnurum* in Kentucky appears to be due to limited suitable habitat in Kentucky. Extensive alluvial streamside deposits (Pomerane 1964) are present from the mouth of Oil Well Branch to about 0.5 km above the mouth of Troublesome Creek, the reach with the largest Kentucky populations of *E. percnurum*. Similar alluvial deposits are almost entirely absent along the remainder of unimpounded portions of Big South Fork in Kentucky. Below the mouth of Bear Creek, suitable habitat continues to decline. Big South Fork narrows and becomes a series of long rapids strewn with large boulders, essentially lacking cobble and small boulder shoals. At Blue Heron, the

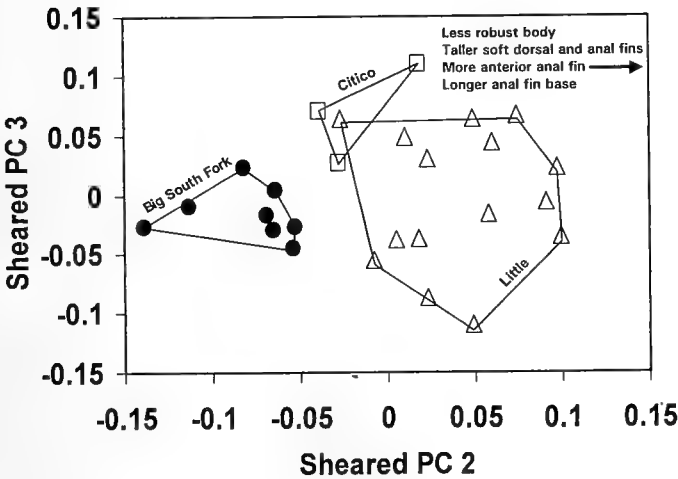


Figure 2. Morphometric scores on sheared PC axes 2 and 3 for 27 *Etheostoma percnurum* from Big South Fork of the Cumberland River, Little River, and Citico Creek, Tennessee.

Table 5. Sheared principal component loadings for 27 morphometric variables for 27 *Etheostoma percnurum* from Big South Fork Cumberland River, Little River, and Citico Creek, Kentucky and Tennessee.

Measurement	Sheared PC 2	Sheared PC 3
Standard length	−0.005	−0.023
Head length	−0.115	0.067
Gape width	0.190	0.303
Pectoral fin length	0.103	0.222
Pelvic fin length	0.144	0.179
First dorsal fin height	−0.162	0.281
Second dorsal fin height	0.392	0.337
Anal fin height at third ray	0.228	0.367
Interorbital width	0.089	−0.099
Snout to occiput	−0.220	0.066
Snout to origin of pelvic fin	−0.044	0.010
First dorsal fin base length	0.143	0.034
Second dorsal fin base length	0.042	−0.161
Pelvic fin origin to anal fin origin	−0.344	0.068
Anal fin base length	0.334	−0.258
Second dorsal fin insertion to hypural	−0.037	0.118
Anal fin insertion to hypural	0.055	−0.290
First dorsal fin origin to anal fin origin	−0.133	−0.012
Pelvic fin origin to second dorsal fin origin	−0.054	0.097
Second dorsal fin origin to anal fin insertion	−0.178	−0.161
Anal fin origin to second dorsal fin insertion	0.296	−0.197
Occiput to pelvic fin origin	−0.025	−0.126
First dorsal fin origin to pelvic fin origin	−0.138	−0.045
Second dorsal fin origin to anal fin origin	−0.007	−0.108
Second dorsal fin insertion to anal fin insertion	0.110	−0.411
Head width	−0.271	0.022
Body width under second dorsal fin origin	−0.352	0.103

river widens, and some suitable habitat is present, although *E. percnurum* was not observed from this area. Below Blue Heron are the impounded waters of Lake Cumberland, certainly unsuitable for *E. percnurum*.

Although the mainstem of Big South Fork is protected from disturbances by the National Park Service, several tributaries (e.g., Bear Creek) are discharging low-quality because of mining in their watersheds. On 26 May 1998, following a rain the previous night, we observed extremely turbid water discharging from Bear Creek into the otherwise clear Big South Fork. Improvement in these impacted streams will help maintain the high-quality habitats in Big South Fork that are required by *E. percnurum* and other species (e.g., *Notropis* sp. “sawfin shiner” and *E. cinereum*) with restricted distributions in Kentucky.

We suggest periodic monitoring of the distribution and abundance of Kentucky *E. percnurum*. We consider both underwater observation and kick-seining around potential rock cover, our primary means of sampling in Big South Fork, as effective and non-lethal. We

recommend that electrofishing not be used to sample this rare species because of the potential harm it can do to fishes (Snyder 1995).

Natural History

Medium to large streams with silt-free rocky pools in the vicinity of riffles seem to be requirements for viable populations of *E. percnurum*. As pointed out by Etnier and Starnes (1993), the habitat of *E. percnurum* is essentially the same as that occupied by *E. cinereum*, a species we found almost invariably associated with *E. percnurum* in Big South Fork. Our general habitat description is similar to the habitat of *E. percnurum* in Little River (Etnier and Starnes 1993) and Copper Creek (Jenkins and Burkhead 1994).

Egg counts in Big South Fork (79–132; mean = 101) were higher than in Little River (23–200; mean = 79; Layman 1991). The higher egg counts per nest in Big South Fork may be attributed to the larger body size of those females. Using the equation of Layman (1991) $\log C = -1.154 + 1.686 \log SL$, females observed in this study would have 51–

61 mature ova, as compared to the 19–44 ova for the smaller females of Little River. Also, nest rock size typically was larger in Big South Fork (mean = 24×19 cm) than in Little River (mean = 16×12 cm; Layman 1991). Larger nest rock size in Big South Fork may simply reflect an abundance of larger nest substrata available. Alternately, this may be a behavioral adaptation in choosing more stable nest rocks in an area with high flows and prone to flash flooding. Additional studies are needed to explore these possibilities. Other nesting biology observations are generally consistent with those of Layman (1991) from Little River, Tennessee and Jenkins and Burkhead (1994) from Copper Creek, Virginia, except Jenkins and Burkhead reported nuptial males from moderate to swift runs.

The heavy infestations and high rate of infection of the black-spot disease observed in this study indicate that it may be an important source of mortality for *E. percunurum*. Although not previously documented for *E. percunurum*, the disease has been reported from many species of North American freshwater fishes. We observed belted kingfishers (*Megaceryle alcyon*) and snails, required intermediate hosts for the strigeid flukes that cause black-spot disease (Berra and Au 1978), to be common in Big South Fork. Heavy infestations have been reported to cause mortalities, particularly during the winter months, in *Esox lucius*, northern pike (Harrison and Hadley 1982), *Lepomis macrochirus*, bluegill (Lemly and Esch 1984), and *Campostoma anomalum*, central stoneroller (Ferrara and Cook 1998). Monitoring of the Big South Fork population of *E. percunurum* should include assessment of the extent of black-spot disease.

Systematics

The differences in squamation, body shape, maximum size, and nesting biology reported here indicate that Big South Fork populations certainly represent an independent evolutionary unit. Considering the relict distribution of this species it seems unlikely that any gene flow has occurred between Cumberland and Tennessee forms in thousands of generations. The morphological variation uncovered in this analysis supports the presence of deep phylogenetic partitions in *E. percunurum*. This pattern suggests that most of the overall diversity

is located among the populations rather than within populations (Meffe and Vrijenhoek 1988). Because the loss of even one of the remaining four populations of *E. percunurum* would cause a substantial loss in diversity of the species, conservation efforts should be directed to preserving as many populations as possible. Protection of the Big South Fork population seems particularly important in maintenance of diversity of *E. percunurum* because of the population's unique morphology and ecology.

ACKNOWLEDGMENTS

Success in finding *E. percunurum* would not have been possible without the assistance of many people and agencies. We are especially appreciative of the underwater expertise of J. R. Shute, P. W. Shute, P. L. Rakes, and K. Harpster (Conservation Fisheries, Inc.). Steve Bakaletz and R. Emmott (National Park Service) provided logistical support, canoes, and river guidance. We thank T. Slone and D. Stephens (Kentucky Department of Fish and Wildlife Resources) and K. M. Cook, D. B. Henry, J. B. Ladonski, K. R. Piller, and J. G. Stewart (Southern Illinois University at Carbondale), K. A. McCafferty, (Morehead State University), and L. V. Eisenhour for assistance in field work. Permits to study *E. percunurum* were provided by Kentucky Department of Fish and Wildlife Resources. R. E. Jenkins, via D. W. Nelson and D. A. Etnier, loaned specimens of *E. percunurum* for comparative purposes.

LITERATURE CITED

- Berra, T. M., and R.-J. Au. 1978. Incidence of black spot disease in fishes in Cedar Fork Creek, Ohio. *Ohio J. Sci.* 78:318–322.
- Biggins, R. E. 1993. Endangered and threatened wildlife and plants: determination of endangered status for the duskytail darter, palezone shiner and pygmy madtom. *Federal Register* 58:25758–25763.
- Biggins, R. E., and P. W. Shute. 1994. Recovery plan for Duskytail Darter (*Etheostoma* [*Catonotus*] sp.). United States Fish and Wildlife Service, Atlanta, GA.
- Bookstein, F. L., B. Chernoff, R. L. Elder, J. M. Humphries Jr., G. R. Smith, and R. E. Strauss. 1985. Morphometrics in evolutionary biology. *Spec. Publ. Acad. Nat. Sci. Philadelphia* 15:1–277.
- Braasch, M. E., and R. L. Mayden. 1985. Review of the subgenus *Catonotus* (Percidae) with descriptions of two new darters of the *Etheostoma squamiceps* species

- group. Occas. Paper Mus. Nat. Hist. Univ. Kansas 119: 1–83.
- Burkhead, N. M., and R. E. Jenkins. 1991. Fishes. Pages 321–409 in K. Terwilliger (coordinator). Virginia's endangered species. McDonald and Woodward Publishing Company, Blacksburg, VA.
- Burr, B. M., and M. L. Warren Jr. 1986. A distributional atlas of Kentucky fishes. Kentucky Nature Preserves Comm. Sci. Techn. Ser. 4.
- Etnier, D. A., and W. C. Starnes. 1993. The fishes of Tennessee. Univ. Tennessee Press, Knoxville, TN.
- Ferrara, A. M., and S. B. Cook. 1998. Comparison of black-spot disease metapopulations in the central stonerollers of two warm-water streams. J. Freshwater Ecol. 13:299–305.
- Harrison, E. J., and W. F. Hadley. 1982. Possible effects of black-spot disease on northern pike. Trans. Am. Fish. Soc. 111:106–109.
- Hubbs, C. L., and K. F. Lagler. 1974. Fishes of the Great Lake region. Univ. Michigan Press, Ann Arbor, MI.
- Humphries, J. M., F. L. Bookstein, B. Chernoff, G. R. Smith, R. L. Elder, and S. G. Poss. 1981. Multivariate discrimination by shape in relation to size. Syst. Zool. 30:291–308.
- Jenkins, R. E., and N. M. Burkhead. 1994. Freshwater fishes of Virginia. American Fisheries Society, Bethesda, MD.
- Jenkins, R. E., and E. A. Lachner. 1971. Criteria for analysis and interpretation of the American fish genera *No-comis* Girard and *Hybopsis* Agassiz. Smithsonian. Contr. Zool. 90:1–15.
- Layman, S. R. 1984. The duskytail darter, *Etheostoma* (*Catonotus*) sp. confirmed as an egg-clusterer. Copeia 1984:992–994.
- Layman, S. R. 1991. Life history of the relict, duskytail darter, *Etheostoma* (*Catonotus*) sp., in Little River, Tennessee. Copeia 1991:471–485.
- Lemly, A. D., and G. W. Esch. 1984. Effects of the trematode *Uvulifer ambloplites* (Hughes, 1927) on juvenile bluegill sunfish, *Lepomis macrochirus*: ecological implications. J. Parasitol. 70:475–492.
- Meffe, G. K., and R. C. Vrijenhoek. 1988. Conservation genetics in the management of desert fishes. Conservation Biol. 2:157–169.
- Page, L. M. 1983. Handbook of darters. Tropical Fish Hobbyist Publications, Neptune City, NJ.
- Page, L. M. 1985. Evolution of reproductive behaviors in percoid fishes. Illinois Nat. Hist. Surv. Bull. 33:275–295.
- Page, L. M., P. A. Ceas, D. L. Swofford, and D. G. Buth. 1992. Evolutionary relationships within the *Etheostoma squamiceps* complex (Percidae; subgenus *Catonotus*) with descriptions of five new species. Copeia 1992:615–646.
- Pomerane, J. B. 1964. Geology of the Barthell Quadrangle and part of the Oneida North Quadrangle, Kentucky. U.S. Geol. Surv. Geol. Quad. Map GQ-314.
- Poss, S. G., and B. B. Collette. 1995. Second survey of fish collections in the United States and Canada. Copeia 1995:48–70.
- SAS Institute. 1985. SAS user's guide: statistics, 1985 edition. SAS Institute, Cary, NC.
- Simon, T. P., and S. R. Layman. 1995. Egg and larval development of the striped fantail darter, *Etheostoma flabellare lineolatum* (Agassiz), and duskytail darter, *E. percnurum* Jenkins, with comments on the *Etheostoma flabellare* species group. Trans. Kentucky Acad. Sci. 56: 28–40.
- Snyder, D. E. 1995. Impacts of electrofishing on fish. Fisheries (Bethesda) 20:26–27.
- Strauss, R. E., and F. L. Bookstein. 1982. The truss: body form reconstruction in morphometrics. Syst. Zool. 31: 113–135.

APPENDIX

Specimens of *E. percnurum* used in morphological comparisons. Museum abbreviations follow Poss and Collette (1995). Parenthetical numbers after catalog numbers refer to the number of specimens used in the meristic and morphometric analyses, respectively.

Big South Fork of the Cumberland River. McCreary County, Kentucky: SIUC 24744 (1,1), SIUC 24761 (1,1), SIUC 24773 (5,5). Scott County, Tennessee: SIUC 24739 (1,1). Copper Creek. Scott County, Virginia: UMMZ 22038 (20,12). Little River. Blount County, Tennessee: UT 91.2719 (7,2), UT 91.2720 (9,4), UT 91.2721 (15,10). Citico Creek. Monroe County, Tennessee: UT 91.2558 (3,3), UT 91.4573 (3,0).

Scientists of Kentucky

David Wendel Yandell, M.D.

Every autumn when the banks along the Ohio blaze in color, the University of Louisville invites to the Falls City one of the nation's renowned surgeons as guest speaker for the annual Yandell Lecture.^a The event celebrates the career of David Yandell (1826–1898), who taught clinical medicine and surgery at the school for more than 30 years. Because he cared passionately about improving medical education, Yandell led a relentless crusade to expand the knowledge of his professional colleagues and to upgrade the quality of care available to their patients.^b

Born near Murfreesboro, Tennessee, Yandell spent his childhood in Lexington, where his father, Lunsford, taught chemistry at Transylvania University. In 1837 the elder Yandell and several of his colleagues founded the Louisville Medical Institute and the family moved to that city. The school attracted students from across the South and throughout the Ohio Valley because of its easy access and its fine faculty, which included Charles Caldwell, Jedediah Cobb, Daniel Drake, Samuel Gross, and Lunsford Yandell. By 1844 when David matriculated, the institute claimed an enrollment of 350.

Despite a growing number of medical schools throughout the nation, few 19th century physicians were well trained. Anyone could call himself a doctor and practice medicine. Most medical men learned their trade by serving a 2-year apprenticeship to a practitioner-preceptor. Some apprentices undoubtedly went along on house calls with their preceptors and thus learned to diagnose and medicate; others probably did little more than drive the doctor's buggy, attend his horse, and sweep out his office. Apprenticeships may have been inferior to medical schools, but no licensing board measured the knowledge of those that took advantage of either—or neither—program.

Throughout most of the century, medical schools had no entrance requirements. They were generally proprietary institutions plagued by feuding faculties, financial handicaps, com-

petition for students, limited curricula, and inadequate teaching aides. To receive a degree, students were required to attend two 4-month sessions (an apprenticeship could be substituted for one session), pass an oral exam, and pay a graduation fee. The exam, apparently the only testing done by the faculty, could be a frightening experience. One of Yandell's classmates compared it to swallowing poison "with no stomach pump about, or [sleeping] with a man with smallpox." He described the examining professors as "dried up specimens of humanity who looked as if they had descended for the occasion from some anatomical museum and who have looked upon death, suffering and the annual ranks of medical aspirants" until their hearts were hard as stone.

Although one of Yandell's professors labeled him a "damned unpromising specimen," he nevertheless passed the exam and graduated from the Louisville Medical Institute in March 1846.^c A few weeks later he sailed to Europe to further his education in the schools of London and Paris. Many of his letters to his family appeared in the *Western Journal of Medicine and Surgery* and the *Louisville Journal*. The cocky Kentuckian greatly admired Sir Robert Lister's surgical techniques and described them in considerable detail, but he sneered at English students who were neither as "intelligent looking" nor as "fine in appearance" as his peers at home. He praised the politeness of French students and found the Parisian professors far more eloquent and interesting than their English brethren. Awed by the teaching hospitals, clinics, and internships available in Paris, Yandell claimed that in a single morning an industrious student could accompany professors through both the medical and surgical wards. And, if "fleet of limb he may follow Roux through his wards at the Hotel Dieu, Jobert through his at St. Louis and hear Velpeau lecture at La Charité!"

By the time he returned to Louisville in autumn 1848, Yandell enjoyed a reputation for excellence among both practitioners and lay-



Figure 1. David Wendel Yandell, ca. 1875.

men. His medications probably were no more successful than those of his colleagues, but his credentials and charm convinced many Louisvillians that he was very knowledgeable. Con-

sequently, medical apprentices as well as patients flocked to him.

One of the problems facing would-be doctors was the lack of a practical method of

studying anatomy and surgical techniques. To aid them—and anyone else who desired the instruction—Yandell and a friend opened a dissection laboratory where they conducted classes in anatomy, physical diagnosis, and surgery. Following his marriage in summer 1851, Yandell closed the facility and moved briefly to Middle Tennessee. However, on his return to Louisville in the mid 1850s, he opened a free outpatient clinic for Louisville's indigents. Modeled after the private clinics of Paris and financed by the fees of medical apprentices and university students, the dispensary supplemented rather than competed with university offerings. Consequently, it and its founder received considerable praise from local practitioners and from doctors who toured it during the 1859 state medical conclave in Louisville.

The success of the dispensary was partially responsible for Yandell being offered a position on the faculty, the realization of a decade-long dream. His major accomplishment as professor of clinical medicine was to talk the faculty into creating three "internships" for the university's top graduates. Unfortunately, the Civil War prevented implementation of the internships.

In summer 1861 Yandell resigned his professorship and received a commission in the Confederate medical department. He spent a few weeks with General Simon Bolivar Buckner's forces at Bowling Green, Kentucky, and then accepted the medical directorship of Albert Sidney Johnston's Army of the West, a command that extended from the Appalachian Mountains to Indian Territory. The task for this military monstrosity would have been a difficult one for a medic with many years of military experience and adequate resources; it was a gargantuan job for a novice. Epidemic diseases, shortage of medical supplies, inadequate hospital facilities, and poorly trained and inexperienced doctors plagued Johnston's army. To care for those felled by measles and other infectious diseases, respiratory ailments, and a variety of illnesses caused by bad food, contaminated water, and inadequate clothing, Yandell commandeered churches, public buildings, and vacant homes across southern Kentucky. When the number of ill exceeded space in the Kentucky facilities, he arranged with the L&N railroad to transport hundreds

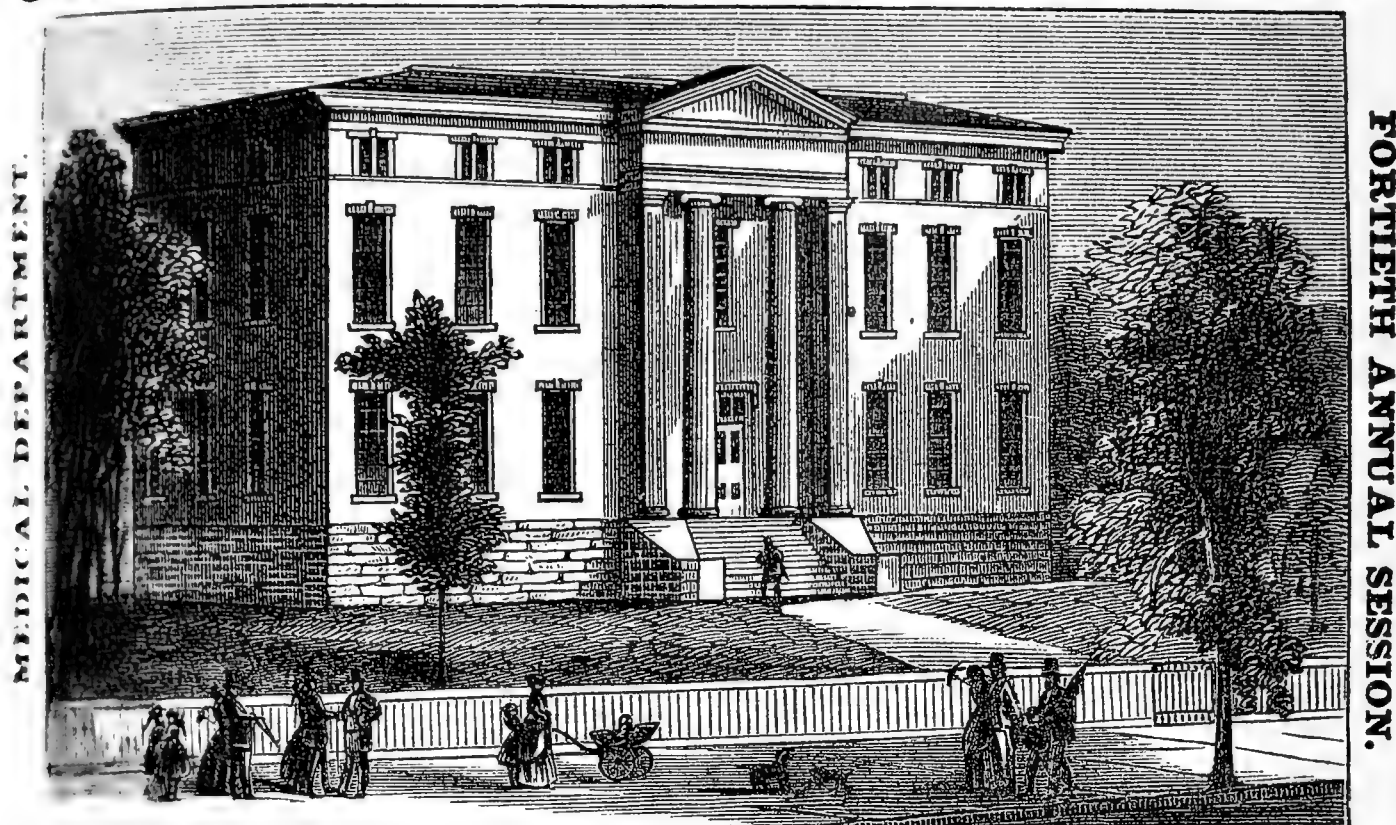
of them to the hospitals and convalescent centers he created in the Nashville area.

Yandell also supervised the employment of medical personnel. Regiments generally selected their own officers, including their regimental physician. Acknowledging the system's fallacy, Yandell created an examining board to screen all medical corps as well as civilian contract applicants. He headed the board and apparently was merciless in his questioning. When asked what he would do for a "shot right through there," the medical director pointing to his own knee, the victim of his grilling about wounds answered, "Well, sir, if it was you that was shot through there, I would not do a d[amne]d thing."

In mid February 1862 the Army of the West retreated into central Tennessee and then to the Corinth, Mississippi, area. In early April the army encountered the enemy near Shiloh Church, about 20 miles north of Corinth. More than 1700 Confederates died during the 2-day Battle of Shiloh; General Albert Sidney Johnston was among them, the victim of a torn popliteal artery. Earlier that morning Yandell had issued tourniquets to all of Johnston's staff, but no one thought to use the lifesaving device. With Johnston's death Yandell lost a friend as well as his coveted position in the Confederate medical department. The Army of the West was renamed and placed under the leadership of General P.G.T. Beauregard, who chose his own medical director. Yandell joined William J. Hardee's corps and traveled with the tactician's forces into Kentucky in autumn 1862. Following the battle of Perryville, he worked day and night administering to the wounded.

In spring 1863 Yandell was sent to Jackson, Mississippi, to "watch over" the health of General Joseph E. Johnston. The two men shared quarters, and the doctor often served as the general's aide, reading aloud his dispatches and writing the replies he dictated. Following the fall of Vicksburg, Yandell analyzed the sad state of affairs in Mississippi that culminated in the river town's loss; he laid the blame on officials in Richmond. His unwise comments reached Confederate president Jefferson Davis. Sensitive to criticism, Davis "banished" Yandell to the Trans Mississippi where the meddling doctor would have "less opportunity for exercising undue influence on the army

UNIVERSITY OF LOUISVILLE.



CORNER OF EIGHTH AND CHESTNUT STREETS.

Figure 2. Medical Department, University of Louisville, Kentucky, ca. 1870.

and community.” Yandell served the remainder of the war as medical director for the army of Edmund Kirby Smith.

Yandell labeled the Civil War a “great though terrible school.” During his 4 years with the army he treated many medical and surgical problems and broadened his understanding of hospital management. He also made valuable contacts and won the admiration of physicians and laymen on both sides. The war’s most profound effect, however, was to sharpen Yandell’s awareness to the nation’s large number of poorly trained and incompetent doctors. To the correction of this shortcoming, he dedicated the remainder of his life.

Returning to Louisville and his medical practice in July 1865, Yandell opened the Louisville Clinic, a large and efficiently organized facility; in 1869 he rejoined the University of Louisville’s medical department. During the war the university had built a small dispensary and contracted two local doctors to operate it. Within a few weeks after Yandell rejoined the faculty, the contract doctors began to complain. They believed that his position on the

faculty gave Yandell’s Louisville Clinic an “immense advantage” over the one they operated. Yandell offered to “divide the influence of his name and services equally between the two dispensaries, to lecture an equal number of times at each and receive no fee from either.” His university colleagues refused his offer and passed a resolution that prohibited professors from teaching private classes. Yandell complained bitterly about the ex post facto ruling and refused to sever his connection with the Louisville clinic. Faculty pressure, however, forced him to rescind his decision.

Because the university’s dispensary was too small to accommodate the students in small groups, much less for class instruction, Yandell began to push for an enlarged university clinic and adjoining amphitheater. Clinical teaching was the “alpha and omega” of a good education, he insisted. By studying medicine “in the laboratory, under the microscope, at the dissection-table, in the wards of the hospital, and in the dispensaries where patients are seen, examined and prescribed for . . . students

learn the diagnosis of disease as well as its treatment," he insisted.

When his colleagues whined that Yandell's building scheme would bankrupt them, he suggested that perhaps the university could convert its academic building, occupied rent-free by Male High School, into a hospital maintained by the city. Indigents could receive free care provided by the faculty and could be studied by students. The scheme would have created one of the first teaching hospitals in the nation. Yandell's colleagues liked the idea but unfortunately the city declined to consider it.

Disappointed but not discouraged, Yandell commenced talks with wealthy Louisvillian Shakespeare Caldwell and convinced him that a hospital was an appropriate memorial to his recently deceased wife. At this facility, Yandell argued, the university's faculty could provide medical and surgical services for impoverished patients and allow students to aid in treating patients and to gain bedside experience. The university offered land near the medical school for the facility. Caldwell agreed to the plan but vetoed the proposed site. Saints Mary and Elizabeth Hospital opened in 1874 but Yandell's plan proved unsuccessful. The distance of several miles between the school and hospital discouraged students from visiting it, and many of the professors could not or would not contribute time beyond their classroom responsibilities to care for patients and supervise students.

Undaunted, Yandell continued his nagging. He suggested that each professor make a contribution and that the faculty thus underwrite the construction of a clinic. The opposition was overwhelming. In frustration, Yandell submitted his resignation with a blistering attack on his colleagues, whom he accused of being disinterested in the quality of medical education. Undoubtedly a few of his harrassed colleagues welcomed Yandell's decision but others knew the university could ill afford to lose its most distinguished professor. Yandell edited the widely read *American Practitioner*, had been elected president of the American Medical Association in 1872, served as its representative to the 1881 International Conference in London, and was one of the founders of the American Surgical Association (and would be its president in 1889). Consequently,

his colleagues promised that if he rescinded his resignation, they would reconsider his plea and try to finance the clinic. Yandell agreed. The university's treasurer cashed city bonds that the school had held for years and the faculty secured a loan for the remainder. H.P. McDonald Brothers received a contract to design and build the clinic and amphitheater. The facility for which Yandell crusaded for nearly 2 decades opened in 1888.

Yandell fought with equal zeal for entrance requirements, an expansion of the curriculum, and a lengthened school year. Unfortunately, competition for students was keen among the city's many medical schools (10 medical schools operated in Louisville during the post-war decades); changes that might increase overhead, raise tuition, or limit admission threatened enrollment and thus the salaries of the proprietary faculty. Yandell's demand for internships likewise met with resistance. The various hospitals were eager to implement the post-graduate positions but wished to make them available to all of the city's medical schools. After years of haggling on how the recipients should be chosen and who would supervise them, the university and the Kentucky Medical School agreed to base the selection on academic merit. Unfortunately, there were only a half-dozen positions for the several hundred graduates eager for the experience.

Yandell's campaign for a more rigorous medical curriculum transcended the commonwealth's borders. In his 1872 presidential address before the delegates to the American Medical Association in Philadelphia, he spoke about the shortcomings of the nation's educational system in general and medical education in particular.

... there are grave defects in the education of many of our students and many of our practitioners of medicine. Not a few of them, I am afraid, have a very slight acquaintance with grammar or physical geography and too many of them know little about etymology and are bad spellers. It is a pity that this is so and I should be glad to see a different state of things."

Answering colleagues who urged the addition of math and Greek to the medical school curriculum, Yandell championed the creation of better primary and secondary schools, not the

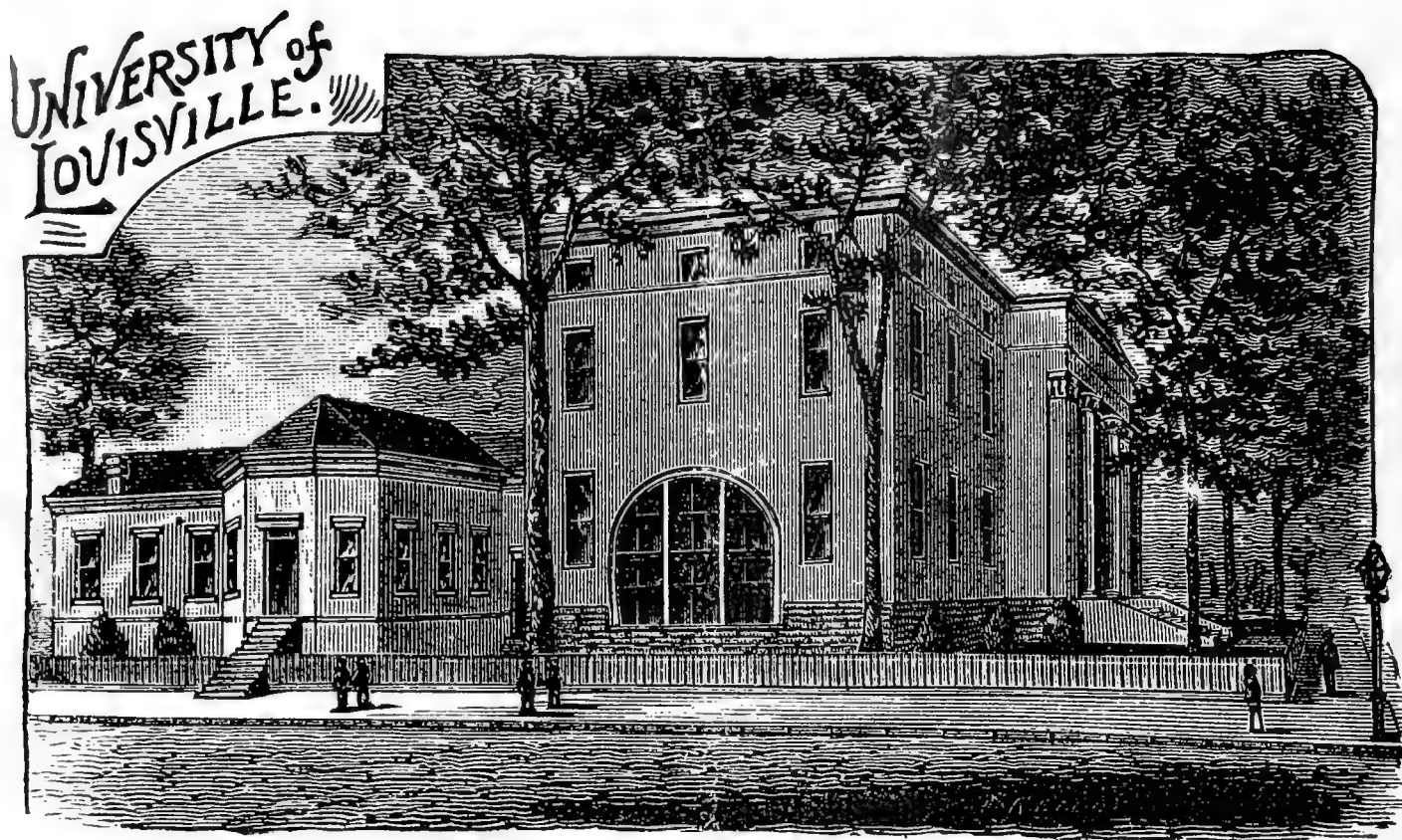


Figure 3. Medical Department, University of Louisville, Kentucky, ca. 1890. The wing on the left is the clinic for which David Yandell fought for 20 years.

teaching of “irrelevant topics” in medical school. Instead, he urged the extension of the lecture term, expansion of the requirements for graduation, increase in the number of professorships and course offering, and more instruction by demonstration. The graduates of the newly inaugurated system at Harvard, he stated, “may know less of Greek and mathematics, [but] are far better trained than they formerly were in clinical medicine and surgery and are better qualified to enter their duties as practitioners.”^d

Although medical education began in the halls of academia, Yandell believed it should continue throughout a doctor’s professional life. For 25 years he was the active senior editor of the *American Practitioner*, a monthly journal that had 2000 subscribers in 1876 and nearly 6000 by 1890. He filled the journal with original articles, reviews of American and European publications, synopses of clinical cases gleaned from other journals, and miscellaneous notes, editorials, and minutes of meetings of local, state, and national medical organizations. Yandell gave his readers a generous and well-balanced feast of medical knowledge and urged his readers to keep current

with developments in the profession. “Life is too short and science too long to permit time to be wasted,” he frequently admonished.

As an classroom teacher Yandell had “few equals and no superiors,” his students recalled, for he enriched their minds, made them wish to learn more, and flavored his lectures with bits of medical history and witty advice. A strong believer in human dignity, Yandell constantly reminded students that indigents deserved the same care and respect as the wealthy. He also urged these future doctors to stay abreast of and keep an open mind to new techniques but to use discretion in adopting them. “If one half of all the certain cures were but certain, the practice of medicine would be too simple to demand special study or require trained followers,” he pointed out.

Yandell also advised that practitioners must know when to act with haste and when to proceed with deliberate caution. He delighted in telling about George IV, whose regular physician was too busy to attend to a small tumor on the royal derrière. Another doctor was called, the growth was removed, and the new doctor was knighted by his grateful sovereign.

The reward might have been excessive, Yandell suggested, but a caring doctor never postponed “until the afternoon . . . messages left for you in the morning.”

In their reminiscences, his former students lauded not only his teaching ability but also wrote of Yandell’s surgical skills. He “cut to the line and to the required depth with geometric precision,” one recalled. For demonstration purposes minor procedures were performed on hospital inmates and dispensary patients but for more difficult operations Yandell used cadavers. Most of the students sat 15 to 30 feet away yet claimed that their teacher’s articulate and detailed explanations compensated for what they could not see. Student memoirs also recalled that Yandell always wore a freshly laundered coat in the “operating” room, scrubbed his hands and instruments with green soap before each procedure, and treated all incisions with compounds of iodine, bromine, and carbolic acid to prevent infection. He advised his students to do likewise. Prior to the general acceptance of antiseptic surgery, such cleanliness was unusual.

Yandell’s admirers included personal friends and private patients, who recalled the bouquets he gave to shut-ins and the groceries he took to indigent families under the tactful guise that the patient needed special foods. Involved in community organizations, he served two terms on the city’s school board, discussed history and literature with fellow members of the Filson and Salmagundi clubs, and founded the Louisville Kennel Club and Louisville Surgical Society. He counted among his close friends attorney Reuben Durrett, editor Henry Watterson, and former Confederates Basil Duke, William Preston, John B. Castleman, and Governor J. Proctor Knott, who appointed Yandell as surgeon general of the Kentucky militia. During one of his trips to Europe to attend a medical conclave, the *British Medical Journal* proclaimed that Yandell was a great favorite in local circles. The Medical Society of London elected him to honorary membership in 1883 and shortly thereafter the London Medical-Chirurgical Society named him an honorary fellow.

When an Englishman referred to him as a Yankee, Yandell protested, assuring that all of his blood “flowed through southern veins.” The staunch Democrat served as one of Ken-

tucky’s hosts during the visits of three Republican presidents—Ulysses S. Grant, Rutherford B. Hayes, and Chester A. Arthur. At an official dinner in Louisville, Yandell entertained Grant with tales of his own Civil War escapades. While accompanying Arthur from Washington to Louisville, a newsman mistook the doctor for the president. Amused by the error, Yandell declared he was the “next best thing” to being president. He was the “Great Presidential Fetcher.”

An articulate lecturer, talented surgeon, dedicated educator, and highly respected citizen, David Yandell was the most progressive and influential member of the university’s medical faculty in the postwar era. In 1887 as part of the organization’s celebration of its hundredth birthday, the College of Physicians and Surgeons of Philadelphia elected 10 American doctors to honorary membership. David Yandell was the only Kentuckian so honored. In spring 1895 the University of Louisville gratefully acknowledged his services when it placed “upon the brow of this our greatest son” the highest degree within its power, the degree of Doctor of Laws.⁶

Nancy Disher Baird
Kentucky Library
Western Kentucky University
Bowling Green, Kentucky 42101

ENDNOTES

- a. Further information about David Yandell is available in: Nancy Disher Baird, *David Wendel Yandell, physician of old Louisville* (Lexington: University Press of Kentucky, 1978). The major sources of information for the book-length biography and this article were the Yandell Family Papers (The Filson Club, Louisville); the Minutes of the Board of Trustees of the University of Louisville (University Archives); the Minutes of the Medical Faculty (Kornhauser Health Sciences Library, Louisville); General and Staff Officers’ File, Old Army Section (National Archives, Washington); Andrew J. Foard Collection (Virginia State Library, Richmond); and Louisville’s newspapers and Yandell’s various published writings and speeches.
- b. Few Kentucky families have had greater influence on medical education than the Yandells of Louisville. Lunsford Pitts Yandell (1798–1878) taught chemistry and pharmacy at Transylvania’s medical department, was one of the founders and teachers of the Louisville Medical Institute (renamed the Medical Department of the University of Louisville in 1846), edited two medical journals, and produced more than 100 sci-



Figure 4. Plaster bust of David Yandell by his niece Enid Yandell. Presented to the University of Louisville ca. 1925.

entific treatises relating to medicine and paleontology. His sons followed in his professional footsteps. William (1844–1901) became a well-known health officer in El Paso and led a campaign to improve sanitation and curb the spread of disease along the Texas-Mex-

ican border. Lunsford Jr. (1837–1885) taught medicine for nearly two decades at the University of Louisville. For additional information about Yandell's family members, see (1) Nancy Disher Baird, "A Kentucky Physician Examines Memphis, Tennessee

- History Quarterly* (autumn 1979), which concerns the early career of David's brother, Lunsford, Jr.; (2) "There Is No Sunday in the Army: The Civil War Letters of Lunsford Pitts Yandell, Jr.," *The Filson Club History Quarterly* (October 1980); (3) "Enid Yandell: Kentucky Sculptor," *The Filson Club History Quarterly* (January 1988). The best-known works by Enid (Lunsford Jr.'s daughter) are the statue of Daniel Boone at the entrance to Louisville's Cherokee Park, Hogan's Fountain inside the park, and the statue of John Thomas in Nashville's Centennial Park. The Filson Club, Kentucky Historical Society, Georgetown College, Speed Museum, and Vanderbilt University (as well as scores of museums, parks, and private collections from Maine to Missouri) also own pieces of her works; and (4) Janet Brockmoller, "Doctor William Martin Yandell," *Password*, 21 (1976). *Password* is the quarterly publication of the El Paso County (Texas) Historical Society.
- c. In spring 1846 the state legislature created the University of Louisville. The "Academical Department" did not materialize until 1907 but the law department opened in autumn 1846 and the Louisville Medical Institute was incorporated as the university's medical department. The transaction for the latter was merely a legal form, for the faculty members continued to elect their own officers, choose new professors (whom the university's trustees automatically approved), collect student fees, and assess themselves when funds were needed for repairs and improvements on their building. In reality the medical school remained autonomous; only its name changed.
 - d. In closing his presidential address, Yandell commented on admitting women to medical school. He could find no satisfactory reason, he said, why women might not succeed "in some line of our profession" for they were "able nurses," but he predicted that a female invasion of the traditional male domain would probably "end in no great results." Certainly he hoped that women would never embarrass the AMA by requesting membership. "I could not vote for that," he promised! Thirteen years later he addressed the graduation class of the short-lived Louisville School of Pharmacy for Women and gave a reason why he believed women might not be competent physicians or pharmacists: "Silence secures accuracy," and women were never quiet, he believed. Wishing the members of his audience good luck, he warned them against mixing a career and marriage, for "if you require your husbands to broil their own chops, you may expect them to wish at least to bray you in one of your own mortars."
 - e. Yandell's last few years were marred by arteriosclerosis, which affected his memory and personality. In 1896 a stroke destroyed his remaining faculties. For nearly 2 years his family nursed the empty shell of a man who once charmed presidents and awed green-horn medical students. Surrounded by those he loved best, David Yandell died 2 May 1898. Two days later a cortege of friends, colleagues, Confederate veterans, and a regiment of state militia conducted his remains to Louisville's Cave Hill Cemetery, where he was buried on a tree-dotted hillside overlooking a picturesque lake.

A Field Checklist of Kentucky Butterflies (Lepidoptera)

Charles V. Covell Jr.

University of Louisville, Louisville, Kentucky 40292-0001

On this and following page is the updated listing of all butterflies known from Kentucky. This supersedes the listing in Covell (1974), and consists of 144 butterfly species, 11 of which are either strays that rarely occur in the Commonwealth, or are known from old records and are not likely to be found in Kentucky now. In my opinion, the known resident and regularly transient butterfly fauna of Kentucky now stands at 133 species.

Scientific names follow Opler (1992), and English (common) names follow Glassberg (ed., 1992). Detailed records and remarks are available in Covell (1999).

I intend for this list to be photocopied by anyone wishing a pocket-sized list of Kentucky butterflies to use in the field. Anyone finding additions or suspected additions to this list is urged to contact me at the above address or at my e-mail address: covell@louisville.edu. I am sure there are a few other butterfly species native to or occasionally established temporarily in Kentucky.

LITERATURE CITED

Covell, C. V., Jr. 1974. A preliminary checklist of the butterflies of Kentucky. J. Lepid. Soc. 28:253-256.
Covell, C. V., Jr. 1999. The butterflies and moths (Lepidoptera) of Kentucky: an annotated checklist. Kentucky State Nature Preserves Comm. Sci. Techn. Ser. 6.
Glassberg, J. (ed). 1995. North American Butterfly Association (NABA) checklist and English names of North American butterflies. North American Butterfly Association, Morristown, NJ.
Opler, P. A. 1998. A field guide to eastern butterflies. Houghton Mifflin, Boston, MA.

FIELD CHECKLIST OF KENTUCKY BUTTERFLIES

by Charles V. Covell Jr., Dept. of Biology, Univ. of Louisville, Louisville, KY 40292-0001 Phone: (502) 852-5942.

Locality _____

Dates _____

Recorded by _____

_____ *Epargyreus clarus* (Silver-spotted Skipper) A

- _____ *Urbanus proteus* (Long-tailed Skipper) S
- _____ *Autochton cellus* (Gold-banded Skipper) U
- _____ *Achalarus lyciades* (Hoary Edge) U
- _____ *Thorybes bathyllus* (Southern Cloudywing) C
- _____ *Thorybes pylades* (Northern Cloudywing) C
- _____ *Thorybes confusus* (Confused Cloudywing) U
- _____ *Staphylus hayhurstii* (Hayhurst's Scallopwing) U
- _____ *Erynnis icelus* (Dreamy Duskywing) C
- _____ *Erynnis brizo* (Sleepy Duskywing) A
- _____ *Erynnis juvenalis* (Juvenal's Duskywing) A
- _____ *Erynnis horatius* (Horace's Duskywing) C
- _____ *Erynnis martialis* (Mottled Duskywing) U
- _____ *Erynnis zarucco* (Zarucco Duskywing) R
- _____ *Erynnis funeralis* (Funereal Duskywing) R
- _____ *Erynnis lucilius* (Columbine Duskywing) R
- _____ *Erynnis baptisiae* (Wild Indigo Duskywing) A
- _____ *Pyrgus centaurae* (Grizzled Skipper) R
- _____ *Pyrgus communis* (Common Checkered Skipper) C
- _____ *Pholisora catullus* (Common Sootywing) C
- _____ *Nastrai lherminier* (Swarthy Skipper) U
- _____ *Lerema accius* (Clouded Skipper) U
- _____ *Ancyloxipha numitor* (Least Skipper) A
- _____ *Thymelicus lineola* (European Skipper) F
- _____ *Hylephila phyleus* (Fiery Skipper) F
- _____ *Hesperia leonardus* (Leonard's Skipper) F
- _____ *Hesperia metea* (Cobweb Skipper) U
- _____ [*Hesperia sassacus* (Indian Skipper)] E
- _____ *Polites peckius* (Peck's Skipper) A
- _____ *Polites themistocles* (Tawny-edged Skipper) A
- _____ *Polites origenes* (Crossline Skipper) F
- _____ *Wallengrenia otho* (Southern Broken-dash) S
- _____ *Wallengrenia egeremet* (Northern Broken-dash) A
- _____ *Pompeius verna* (Little Glassywing) U
- _____ *Atalopedes campestris* (Sachem) A
- _____ *Anatrytone logan* (Delaware Skipper) U
- _____ *Poanes hobomok* (Hobomok Skipper) F
- _____ *Poanes zabulon* (Zabulon Skipper) C
- _____ *Poanes yehl* (Yehl Skipper) C
- _____ *Poanes viator* (Broad-winged Skipper) R
- _____ *Euphyes dion* (Dion Skipper) F
- _____ *Euphyes dukesi* (Duke's Skipper) R
- _____ *Euphyes vestris* (Dun Skipper) C
- _____ *Atrytonopsis hianna* (Dusted Skipper) U
- _____ *Amblyscirtes hegon* (Pepper and Salt Skipper) U
- _____ *Amblyscirtes aesculapius* (Lace-winged Roadside Skipper) U
- _____ *Amblyscirtes vialis* (Common Roadside Skipper) U
- _____ *Amblyscirtes belli* (Bell's Roadside Skipper) U
- _____ *Lerodea eufala* (Eufala Skipper) R
- _____ *Panoquina ocola* (Ocola Skipper) U
- _____ *Battus philenor* (Pipevine Swallowtail) A

- *Battus polydamas* (Polydamas Swallowtail)] S
 ——— *Eurytides marcellus* (Zebra Swallowtail) C
 ——— *Papilio polyxenes asterius* (Black Swallowtail) C
 ——— *Papilio joanae* (Joan's Swallowtail) R
 ——— *Papilio cresphontes* (Giant Swallowtail) F
 ——— *Papilio glaucus* (Tiger Swallowtail) C
 ——— *Papilio troilus* (Spicebush Swallowtail) C
 ——— *Papilio palamedes* (Palamedes Swallowtail) R
 ——— *Pontia protodice* (Checkered White) U
 ——— *Pieris virginensis* (West Virginia White) C
 ——— *Pieris rapae* (Cabbage White) C
 ——— *Euchloe olympia* (Olympia Marble) U
 ——— *Anthocharis midea* (Falcate Orange Tip) C
 ——— *Colias philodice* (Clouded Sulphur) A
 ——— *Colias eurytheme* (Orange Sulphur) A
 ——— *Colias cesonia* (Southern Dogface) U
 ——— *Phoebis sennae* (Cloudless Sulphur) C
 ——— [*Phoebis philea* (Orange-barred Sulphur)] S
 ——— [*Phoebis agarithe* (Large Orange Sulphur)] S
 ——— [*Kricogonia lyside* (Lyside Sulphur)] S
 ——— *Eurema lisa* (Little Yellow) F
 ——— *Eurema nicippe* (Sleepy Orange) F
 ——— *Nathalis iole* (Dainty Sulphur) U
 ——— *Feniseca tarquinius* (Harvester) U
 ——— *Lycaena phlaeas americana* (American Copper)
 ——— *Lycaena hyllus* (Bronze Copper) F
 ——— *Atlides halesus* (Great Purple Hairstreak) R
 ——— *Satyrium titus mopsus* (Coral Hairstreak) C
 ——— [*Satyrium acadicum* (Acadian Hairstreak)] S
 ——— *Satyrium edwardsii* (Edwards' Hairstreak) C
 ——— *Satyrium calanus falacer* (Banded Hairstreak) C
 ——— *Satyrium caryaevorum* (Hickory Hairstreak) U
 ——— *Satyrium liparops* (Striped Hairstreak) U
 ——— *Satyrium favonius ontario* (Northern Hairstreak) U
 ——— *Callophrys grynea* (Juniper Hairstreak) C
 ——— *Callophrys augustinus* (Brown Elfin) U
 ——— *Callophrys irus* (Frosted Elfin) R
 ——— *Callophrys henrici* (Henry's Elfin) C
 ——— *Callophrys niphon* (Eastern Pine Elfin) U
 ——— *Parrhasius m-album* (White-M Hairstreak) R
 ——— *Erora laetus* (Early Hairstreak) R
 ——— *Calycopis cecrops* (Red-banded Hairstreak) U
 ——— *Strymon melinus* (Gray Hairstreak) C
 ——— *Leptotes marina* (Marine Blue) R
 ——— *Everes comyntas* (Eastern Tailed Blue) A
 ——— *Celastrina argiolus ladon* (Spring Azure) C
 ——— *Celastrina ebenina* (Dusky Azure) C
 ——— *Celastrina neglectamajor* (Appalachian Blue) U
 ——— *Glaucopsyche lygdamus* (Silvery Blue) U
 ——— *Calephelis borealis* (Northern Metalmark) U
 ——— *Calephelis mutica* (Swamp Metalmark) R
 ——— *Libythea carinenta bachmanii* (American Snout Butterfly) C
 ——— *Agraulis vanillae* (Gulf Fritillary) U
 ——— *Euptoietia claudia* (Variegated Fritillary) C
 ——— *Speyeria diana* (Diana Fritillary) C
 ——— *Speyeria cybele* (Great-spangled Fritillary) A
 ——— *Speyeria aphrodite* (Aphrodite Fritillary) U
 ——— *Speyeria idalia* (Regal Fritillary) E
 ——— *Boloria bellona* (Meadow Fritillary) C
 ——— [*Boloria selene myrina* (Silver-bordered Fritillary)] S
 ——— *Chlosyne gorgone* (Gorgone Checkerspot) R
 ——— *Charidryas nycteis* (Silvery Checkerspot) C
 ——— *Phyciodes tharos* (Pearl Crescent) A
 ——— *Phyciodes batesii* (Tawny Crescent)] R
 ——— *Euphydryas phaeton* (Baltimore Checkerspot) U
 ——— *Polygonia interrogationis* (Question Mark) C
 ——— *Polygonia comma* (Eastern Comma) C
 ——— *Polygonia faunus smythi* (Green Comma) R
 ——— *Polygonia progne* (Gray Comma) R
 ——— [*Nymphalis vau-album j-album* (Compton Tortoise Shell)] S
 ——— *Nymphalis antiopa* (Mourning Cloak) F
 ——— *Aglais milberti* (Milbert's Tortoise Shell) S
 ——— *Vanessa virginiensis* (American Lady) U
 ——— *Vanessa cardui* (Painted Lady) C
 ——— *Vanessa atalanta* (Red Admiral) C
 ——— *Junonia coenia* (Common Buckeye) C
 ——— *Anartia jatrophae* (White Peacock) S
 ——— *Limenitis arthemis arthemis* (White Admiral) S
 ——— *Limenitis arthemis astyanax* (Red Spotted Purple) C
 ——— *Limenitis archippus* (Viceroy) C (KY'S STATE BUTTERFLY)
 ——— *Anaea andria* (Goatweed Leafwing) U
 ——— *Asterocampa celtis* (Hackberry Emperor) C
 ——— *Asterocampa clyton* (Tawny Emperor) C
 ——— *Enodia portlandia missarkae* (Southern Pearly-Eye) U
 ——— *Enodia anthedon* (Northern Pearly-Eye) C
 ——— *Enodia creola* (Creole Pearly-Eye) U
 ——— *Satyroides appalachia* (Appalachian Brown) C
 ——— *Cyllopsis gemma* (Gemma Satyr) U
 ——— *Hermeuptychia sosybius* (Carolina Satyr) U
 ——— *Megisto cymela* (Little Wood Satyr) C
 ——— *Cercyonis pegala* (Common Wood-Nymph) C
 ——— *Danaus plexippus* (Monarch) C
 ——— [*Danaus gilippus* (Queen)] S

Additions: _____

[brackets indicate questionable specimens or sight records for Ky.]

A = Abundant; C = Common; F = Frequent; U = Uncommon; R = Rare; E = probably endangered or extirpated; S = stray, not native to Kentucky.

Notes on North American *Elymus* Species (Poaceae) with Paired Spikelets: I. *E. macgregorii* sp. nov. and *E. glaucus* ssp. *mackenzii* comb. nov.

Julian J.N. Campbell

The Nature Conservancy (Kentucky Chapter), 642 West Main Street, Lexington, Kentucky 40508

ABSTRACT

Elymus macgregorii R. Brooks & J.J.N. Campbell, sp. nov., is here described. Though widespread in eastern North America, it has been generally confused with *E. virginicus* and *E. glaberrimus*. It has a more open spike, with long awns, and there are other slight differences. It flowers about a month earlier and occurs in woodlands on relatively mesic, fertile soils. The new combination *E. glaucus* Buckley ssp. *mackenzii* (Bush) J.J.N. Campbell is provided for plants of rocky calcareous glades in the Ozark-Ouachita region, disjunct by 800 km from the main range of *E. glaucus* in western North America. Compared to other subspecies, ssp. *mackenzii* usually has narrower, pubescent leaf blades and longer glume awns.

INTRODUCTION

Elymus L. has been one of the most difficult genera of North American grasses to understand taxonomically. Published treatments range from simplistic (e.g., Gould 1975) to intricate (e.g., Bowden 1964). *Elymus virginicus* L. and closely allied taxa—a group characterized by relatively large, thick, basally indurate, bowed-out glumes that disarticulate from erect spikes—have been especially troublesome, although improvements in their treatment were advanced by the master's thesis of Brooks (1974). Variation within *E. glaucus* Buckley, a large complex species, also remains poorly understood (Snyder 1950, 1951; Stebbins 1957). I am currently completing a treatment of North American *Elymus* species with paired spikelets for the new Manual of North American Grasses (M.E. Barkworth, K.M. Kapels, and L.A. Vorobik, in preparation), which has already necessitated some taxonomic notes (Campbell 1995, 1996). This paper continues by (1) describing the following new species related to *E. virginicus* and (2) by providing a new subspecific combination in *E. glaucus*.

1. *ELYMUS MACGREGORII* sp. nov.

In the late 20th century, it is rare to recognize a new species of vascular plant that is widespread in eastern North America. Nevertheless, based on much morphological and phenological study, Brooks (1974) showed that the plants described below are distinct. He initially treated them as *Elymus virginicus*

var. *minor* Vasey ex L.H. Dewey (1892, p. 550), but later (in McGregor et al. 1986, p. 1171) he noted that further study was needed. The type of *E. virginicus* var. *minor* has proven difficult to interpret: collected in “northern Texas” [without date] by [S.B.] Buckley s.n. (US 1020445), it consists of just one spike with an upper culm section and a few leaves. Its rachis internode lengths and awn lengths suggest that this specimen may be transitional between the species described below and *E. virginicus* var. *jejunus* (Ramaley) Bush. Moreover, “minor” cannot be used as a new specific epithet in this genus because it has already been used for a different species, as *E. minor* (J.G. Smith) M.E. Jones (1912), a name based on *Sitanion minus* J.G. Smith, which is now an accepted synonym of *E. elymoides* (Raf.) Swezey [= *S. hystrix* (Nutt.) J.G. Smith.].

Elymus macgregorii R. Brooks & J.J.N. Campbell, sp. nov. Figure 1a.

Plantae caespitosae, plerumque glaucopruinosae. Culmi 40–120 cm longi, erecti vel leviter decumbentes; nodi plerumque nudi. Foliorum vaginae glabrae vel raro villosae; ligulae minus quam 1 mm; auriculae 2–3 mm, purpurascens vel nigrescens; laminae 7–15 mm latae, laxae, supra glabrae vel interdum villosae, nitido-atrovirides sub pruina pallidoglaucæ. Spicae 4–12 cm longae, 2.5–4 cm latae, erectae, exsertae; nodi 9–18, unusquisque 2(3) spiculis; internodiae 4–7 mm longae, tenuiae (sectionibus angustissimis ca. 0.3

mm), sine angulis dorsalis prominentis. Spiculae 10–15 mm longae (minus aristae), effusae, glaucae vel maturite stramineae-fuscae, cum (2)3–4 flosculis, flosculus infirmus cadens cum glumae et basa rachillae affixae. Glumae 8–16 mm longae, 1–1.8 mm latae, basalia 1–3 mm induratae (nervis obscuris) et moderate exarcuratae, corpe lineari-lanceolato, plerumque glabro vel scabro, venis (2)4–5(8), arista 10–25 mm longa, stricta vel interdum contorta in spiculae infimae; lemmae 6–12 mm longae, plerumque glabrae vel scabrae vel interdum villosae ad hirsutae, arista (15)20–30 mm, stricta; paleae 6–10 mm longae, obtusae; antherae 2–4 mm longae, plerumque manifestae e mensis quintus serotinus ad mensis sextus medius; chromosomatum numerum, $2n = 28$.

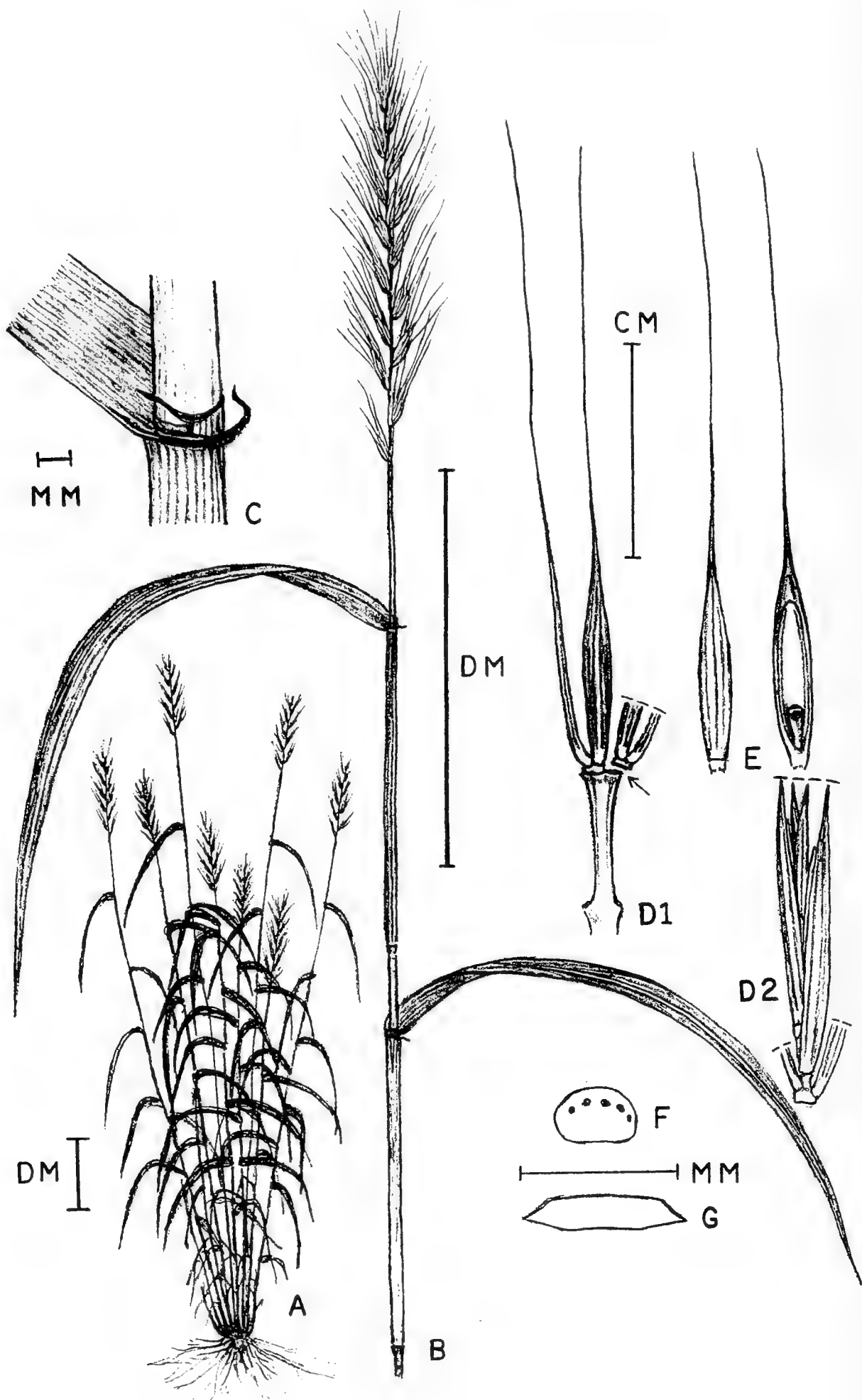
Plants caespitose, usually glaucous-pruinose. Culms 40–120 cm, erect or slightly decumbent; nodes mostly exposed. Leaf sheaths glabrous or rarely villous; ligules under 1 mm; auricles 2–3 mm, purplish to black; blades 7–15 mm wide, lax, glabrous or occasionally villous above, dark glossy green under the pale glaucous waxy bloom. Spikes 4–12 cm long, 2.5–4 cm wide including awns, erect, exserted; nodes 9–18, each with 2(3) spikelets; internodes 4–7 mm, thin (narrowest section ca. 0.3 mm), without prominent dorsal angles. Spikelets 10–15 mm (minus awns), spreading, glaucous then maturing to pale yellowish brown, with (2)3–4 florets, the lowest floret disarticulating with glumes and rachilla base attached. Glumes subequal, 8–16 mm long, 1–1.8 mm wide, the basal 1–3 mm indurate (with veins hidden) and moderately bowed out, the body linear-lanceolate, usually glabrous or scabrous, (2)4–5(8)-veined, the awn 10–25 mm, straight or occasionally contorted in the lowest spikelets; lemmas 6–12 mm, usually glabrous or scabrous, occasionally villous to hirsute, the awn (15)20–30 mm, straight; paleas 6–10 mm, obtuse; anthers 2–4 mm, usually evident from mid-May to mid-June. Chromosome number, $2n = 28$ (Brooks 1974).

TYPE: U.S.A., KENTUCKY, Fayette Co., wooded banks of West Hickman Creek near Armstrong Mill Road, 31 May 1998, *J. Campbell* 98-001 (HOLOTYPE: US; ISOTYPES: KY, KANU, KNK, MADI, MO, NCU, WIS).

I have annotated many collections at

KANU, KNK, KY, ISC, MADI, MO, NCU, OKL, TEX, UARK, US, UTC, VDB, WIS, and elsewhere with the above name or with the earlier suggested names *E. virginicus* var. *minor* and “*E. interior*” ined. A distribution map is provided in Figure 1b, and recorded counties are listed in the Appendix. *Elymus macgregorii* occurs mostly in the Mississippi River and Ohio River drainages (Texas to South Dakota, Alabama to Ohio), but it also extends eastward to the Piedmont and New England (North Carolina to Maine), and westward to central Texas (Figure 1b). It has not yet been confirmed from Canada, but there is a possible atypical specimen from Nova Scotia (see Appendix), and the illustration of “*Elymus hystrix* L.” in Dore and McNeill (1980) appears to be *E. macgregorii*, based on the long awns, distinct glumes, and upward-pointing spikelets. Its typical habitats are in mesic woodlands and thickets on fertile alluvium or, in a few regions (such as the “Bluegrass” of Kentucky, Indiana, and Ohio), on unusually fertile, base-rich upland residuum. In range and habitat, this species is somewhat similar to two widespread oaks (Little 1971), *Quercus macrocarpa* Michx. (excluding *Q. mandanensis* Rydb.) and *Q. muhlenbergii* Engelm. (excluding *Q. prinoides* Willd.).

Elymus macgregorii has been overlooked largely because its morphological distinctions are often not obvious at first inspection, especially in the herbarium. Yet, in 20 years of experience with this species in Kentucky, near the center of its range, I have found that, in addition to having some slight visible differences, it consistently flowers about a month earlier than its closest relatives, *E. glabriflorus* (Vasey ex L.H. Dewey) Scribn. & C.R. Ball and *E. virginicus*, including var. *jejunus* (Ramaley) Bush and var. *intermedius* (Vasey ex A. Gray) Bush. By recording dates of anthesis in the field and herbarium, I have confirmed the results of Brooks’s (1974) garden studies that first demonstrated this phenological difference. I have also observed that the habitat of *E. macgregorii* is more restricted to woodlands on highly fertile, mesic soils, which, due to their productivity, are prone to much biotic disturbance. For example, this species appears to have been particularly abundant before settlement, along with the globally threatened



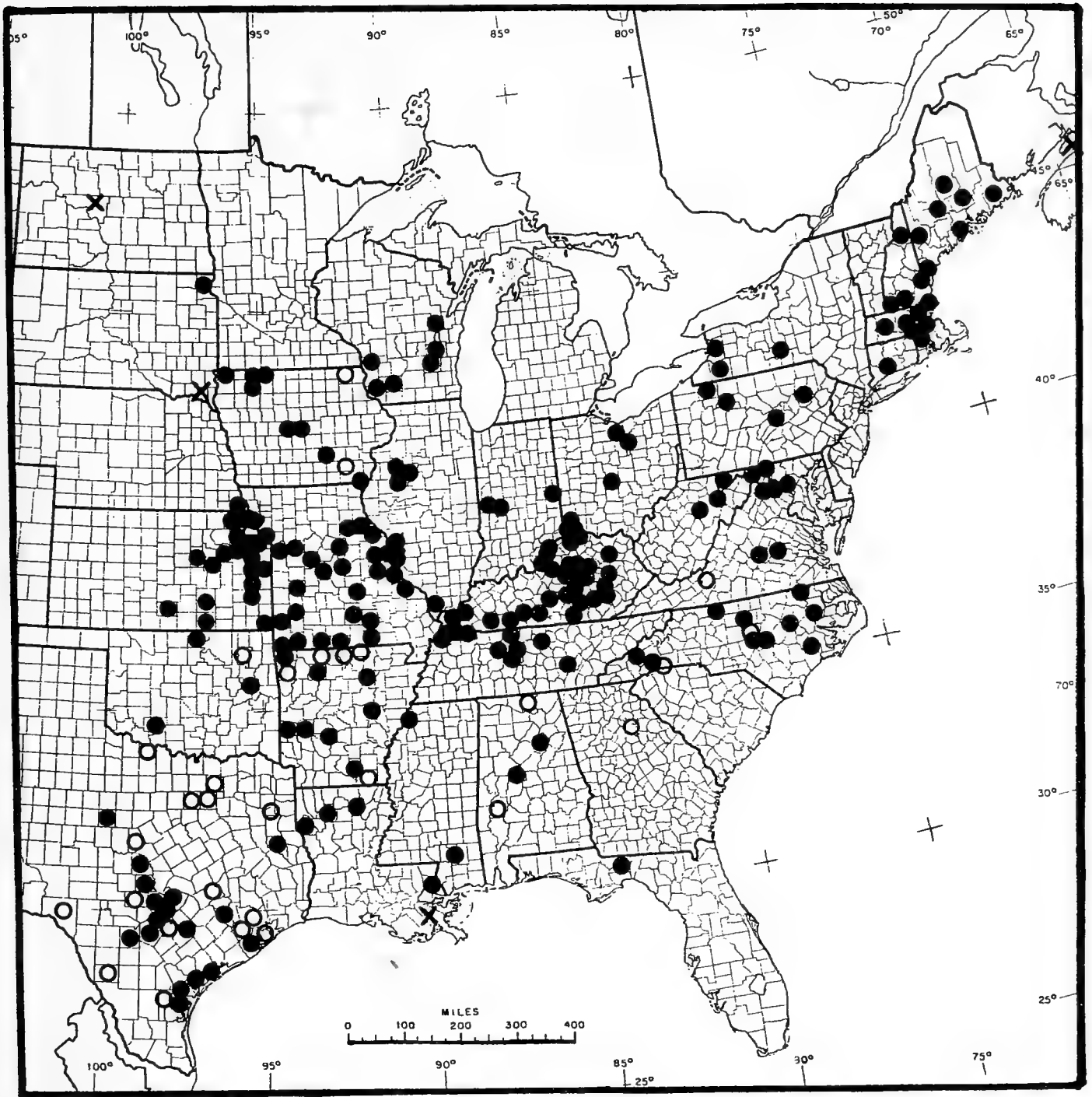


Figure 1b. *Elymus macgregorii*: mapped county records (eastern North America). Solid dots show counties recorded with typical plants; open dots show counties recorded with only atypical plants that may be transitional to *E. virginicus* var. *jejunus* (see text); crosses show counties with other atypical plants of uncertain identification, which may be hybrids in some cases. This map is based on herbarium specimens examined by the author (see Appendix) (a complete search of all major state herbaria has not yet been made).

←

Figure 1a. *Elymus macgregorii* R. Brooks & J.J.N. Campbell.—A. Habit.—B. Upper portion of culm with mature spike, viewed on plane with alternating spread of spikelets.—C. Sheath summit and blade base.—D1. Mature rachis internode and glumes, viewed in plane of spikelet spread (with abaxial view of central glume in spikelet, and largely side view of lateral glume); arrow indicates disarticulation of right-hand glume pair and attached rachilla base from rachis.—D2. Spikelet, with lateral view of florets.—E. Mature floret in abaxial view (left) and adaxial view (right); note rounded palea summit shorter than lemma body.—F. Cross-section of mature, indurate glume base, showing embedded vascular bundles on abaxial side.—G. Cross-section of central rachis internode, showing only slight angles on abaxial side. Drawn from unpressed robust material in the same population as the holotype.

Trifolium stoloniferum Muhl., in the open, ungulate-browsed woodlands of Kentucky's Inner Bluegrass plains (Campbell et al. 1988). Such vegetation is now virtually all cleared for farmland, and *E. macgregorii* survives only in forested stream corridors, woodlots, and fencerows without frequent grazing or mowing.

Elymus macgregorii has often been confused with *E. glabriflorus* [syn. *E. virginicus* var. *glabriflorus* (Vasey ex L.H. Dewey) Bush]. The latter is a species of the southeastern United States, typical of native grasslands and open woodlands on subhydric to subxeric sites that need not have particularly high soil fertility. *Elymus macgregorii* usually sheds pollen and begins to set seeds in mid-May to mid-June, as compared with mid-June to late July in *E. glabriflorus*. It has a more open spike with longer, more exposed rachis internodes ca. 4–7 mm, compared with 3–5 mm in *E. glabriflorus*. The spikes are often shorter and typically have about 9–18 nodes; in contrast, spikes of *E. glabriflorus* typically have 15–30 nodes, unless unusually stunted. In many cases, there may be little or no obvious vegetative differences. However, in fresh condition the leaves of *E. macgregorii* are generally lax and dark, glossy green under the distinct, pale, glaucous, waxy bloom, whereas those of *E. glabriflorus* range from lax (especially in shade) to ascending and somewhat involute (especially in sun), and they are generally paler, duller green with or without a waxy bloom. The auricles of *E. macgregorii* are typically prominent, ca. 2–3 mm, and turn from purplish to black at maturity, whereas those of *E. glabriflorus* (and *E. virginicus*) are typically less developed, ca. 0–2 mm, and only brownish at maturity.

Both *Elymus macgregorii* and *E. glabriflorus* can be distinguished from *E. virginicus* by their fully exserted, wide spikes (ca. 2.5–6 cm wide including awns), with more spreading spikelets and longer lemma awns (ca. 15–40 mm). Spikes of *E. virginicus* vary from fully exserted to remaining partly enclosed by the upper leaf sheaths and are only 0.7–2 cm wide; its spikelets are appressed to slightly spreading, and lemma awns are 1–15(20) mm long. While no consistent vegetative differences have been found, *E. macgregorii* and *E. glabriflorus* are sometimes pubescent in spikes

(especially lemmas) and leaves (sheaths and upper blade surfaces), whereas *E. virginicus* is generally glabrous to scabridulous except for a few varieties or forms that have rather narrow ranges in distribution or habitat. A later paper in this series will present a detailed key to these species and other *Elymus* spp. with paired spikelets.

Variation within *Elymus macgregorii* deserves further study. Throughout much of its range, plants with pubescent spikes (especially lemmas) occur at scattered locations, but these have not yet been reported in distinct habitats or in large enough populations to warrant taxonomic recognition. However, some plants from Missouri, Arkansas, Oklahoma, and Texas, including the type of *E. virginicus* var. *minor*, do deserve more detailed study for possible recognition. These have smaller spike dimensions (with internodes down to 3–4 mm and awns down to 10 mm), and often less glaucous foliage. They may represent a transition to *E. virginicus* var. *jejunus*, though often with distinctly villous leaves. Thus, separation of *E. macgregorii* from *E. virginicus* var. *jejunus* may remain difficult in this region without further research.

My examination of herbarium material suggests that *Elymus macgregorii* forms rare natural hybrids with *E. virginicus*, *E. glabriflorus*, *E. hystrix* L., and perhaps *E. villosus* Muhl. ex Willd. Also, while misidentified as *E. virginicus* var. *intermedius* (Vasey ex A. Gray) Bush, *E. macgregorii* appears to have been artificially hybridized by Stebbins and Snyder (1956) with *E. glaucus*, *E. stebbinsi* Gould, and *Pseudoroegneria spicata* (Pursh) A. Löve [syn. *Agropyron spicatum* Pursh]. This misidentification is indicated by examination of their probable voucher material at US (G.L. Church s.n., Limington, Vermont), and by its attributed characters in their Table 2: rachis internodes ca. 7 mm, glumes ca. 28 mm, and lemmas ca. 28 mm (including awns).

Elymus macgregorii is named in honor of the Clan M[a]cGregor, as represented by two descendants: (1) Ronald Leighton McGregor, emeritus professor and former herbarium director at the University of Kansas (Lawrence), Great Plains botanist (McGregor et al. 1986), and supervisor of Brooks (1974); and (2) John MacGregor (of Nicholasville, Kentucky), an outstanding Kentucky naturalist, explorer of

Inner Bluegrass thickets and other disturbed places, and my occasional collaborator (e.g., Campbell et al. 1994). An appropriate English name is “early wild-rye” since this is the first species of *Elymus* to flower in east-central North America.

2. *ELYMUS GLAUCUS* ssp. *MACKENZII*
comb. nov.

Elymus glaucus ssp. *glaucus* is a widespread taxon in western North America (Figure 2b). There are two other described subspecies, which are confined to regions along the Pacific Coast and adjacent mountain ranges, and there is much additional variation that deserves further attention (Snyder 1950, 1951; Stebbins 1957). For example, some plants can have solitary spikelets at most nodes, often resembling *E. trachycaulus* (Link) Gould ex Shinners according to M.E. Barkworth (Utah State University, pers. comm., June 1997). The main range of *Elymus glaucus* extends east to the western Great Plains, but this species is unknown or very rare in most of Texas, Oklahoma, Kansas, and Nebraska. Its northern range extends east to a few scattered records in the Great Lakes region (e.g., Voss 1972). Some Great Lakes records, however, are questionable due to possible confusion with *E. trachycaulus*, or possible post-Columbian spread of *E. glaucus* to the east.

This section of the paper is concerned with the remarkably disjunct southeastern segregate of this species in the Ozark Mountains and Ouachita Mountains of eastern Oklahoma, northwestern Arkansas, and southwestern Missouri. These plants occur mostly in rocky, calcareous, xeric openings. They were originally described as *Elymus mackenzii* Bush (1926), based on Missouri specimens, but no subsequent author has recognized them as distinct from *E. glaucus*. The taxon is resurrected here as a subspecies of *E. glaucus* because of its slight-morphological differences and its great disjunction of about 800 km from the nearest records of *E. glaucus* to the west and north.

Elymus glaucus Buckley ssp. *mackenzii*
(Bush) J.J.N. Campbell, comb. nov. Figure 2a.

Basionym: *Elymus mackenzii* Bush, Am. Midl. Naturalist 10:53, 1926.

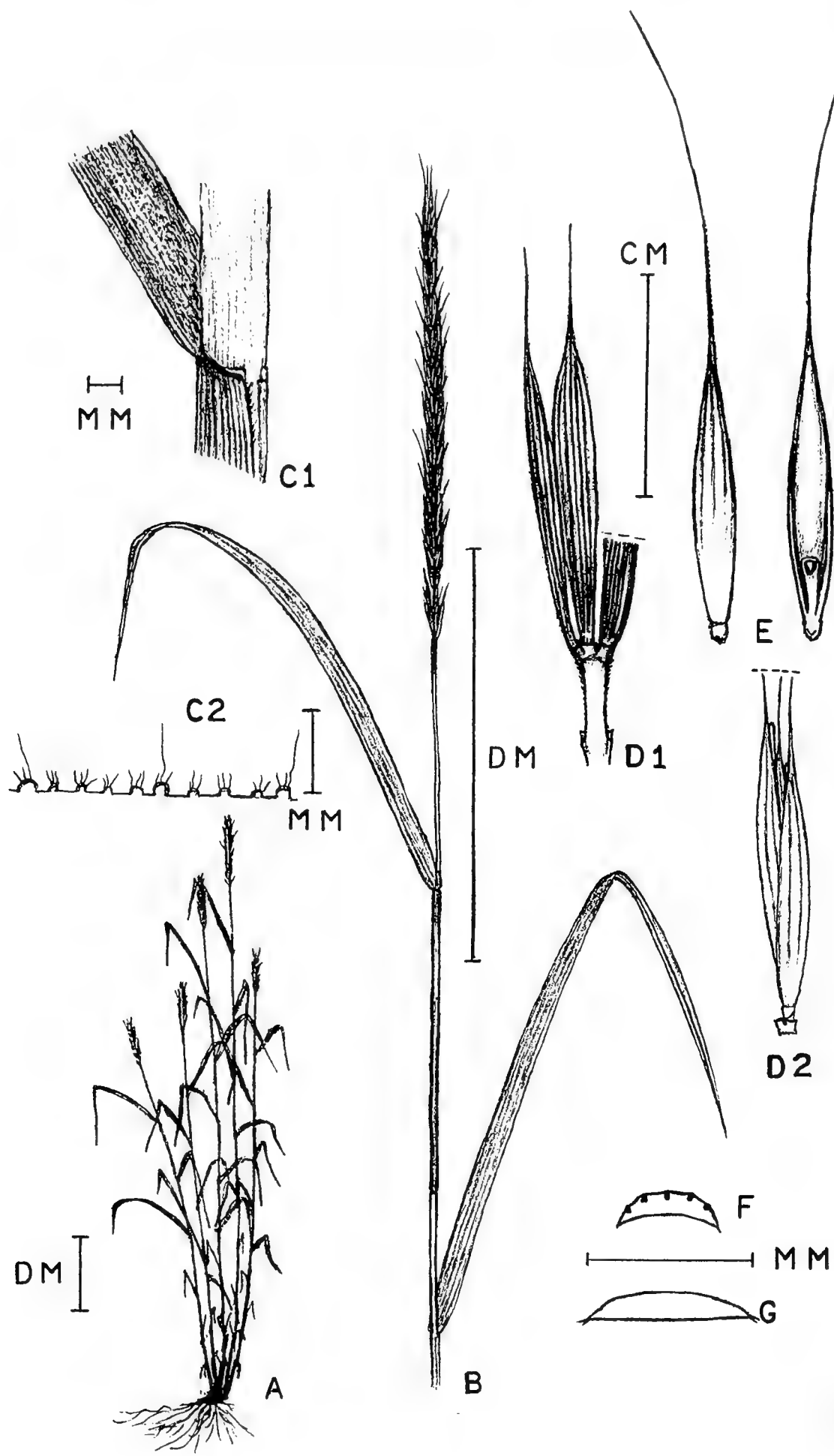
TYPE: U.S.A., MISSOURI, [Barry Co.] Eagle Rock, 15 Jun 1897, *B.F. Bush* 77 (HOLOTYPE: US 318128!).

OTHER COLLECTIONS EXAMINED. U.S.A., ARKANSAS: Carroll Co., wooded northeast facing slope along White River at Catron Bend, 5 miles northwest of Eureka Springs, 27 Jul 1953, *M.J. May* 12 (UARK); Polk Co., Rich Mountain, margin of rich woods, 30 Jun 1967, *G.E. Tucker* 5439 (APCR, NCU). MISSOURI: Barry Co., 3 miles east of Roaring Rv. State Park on high limestone *Juniperus* glade, 10 miles southeast of Cassville, *H.H. Iltis et al.* 1, 3 Jul 1960 (WIS, KY). OKLAHOMA: Le Flore Co., summit of Rich Mt. near Arkansas line, damp ledge, rock in full sun, 24 Jun 1980, *R. Kral* 65492 (VDB).

Another Missouri record is from Moore (1954), who listed “*Elymus mackenzii*” from Stone Co. on xeric limestone bluffs along the White River. I have not yet located voucher collections by Moore, but H.H. Iltis (Univ. of Wisconsin, pers. comm., Nov 1996) worked with him and has confirmed the identification. Also, Steyermark (1963) mapped *E. glaucus* from eight counties in southwestern Missouri (Barry, Barton, Jasper, Lawrence, Newton, Ozark, Stone, and Taney). His collections, mostly at UMO (and perhaps F), have been partly examined by Yatskievych (1999), whose description of *E. glaucus* suggests that all Missouri plants belong to ssp. *mackenzii*, except perhaps for a old collection from Jackson Co. with unknown location and affinity: 21 Jul 1892, *B.F. Bush* 2849 (UMO). Steyermark (1963) noted that the species is “usually found on rocky limestone ledges of bluffs along White River and tributaries and other streams.”

The following key distinguishes the described subspecies of *Elymus glaucus*. However, although ssp. *jepsonii* was recognized in Hickman (1993), this taxon may not deserve subspecies status according to M.E. Barkworth (pers. comm., April 1999). A complete revision of this complex species is needed.

1. Lemma awns (0)1–5(7) mm; glume awns 0–2 mm; blades glabrous to scabrid-puberulent above
..... ssp. *virescens* (Piper) Gould
1. Lemma awns (5)10–25(35) mm; glume awns (0.5)1–8(9) mm; blades glabrous or variously pubescent.



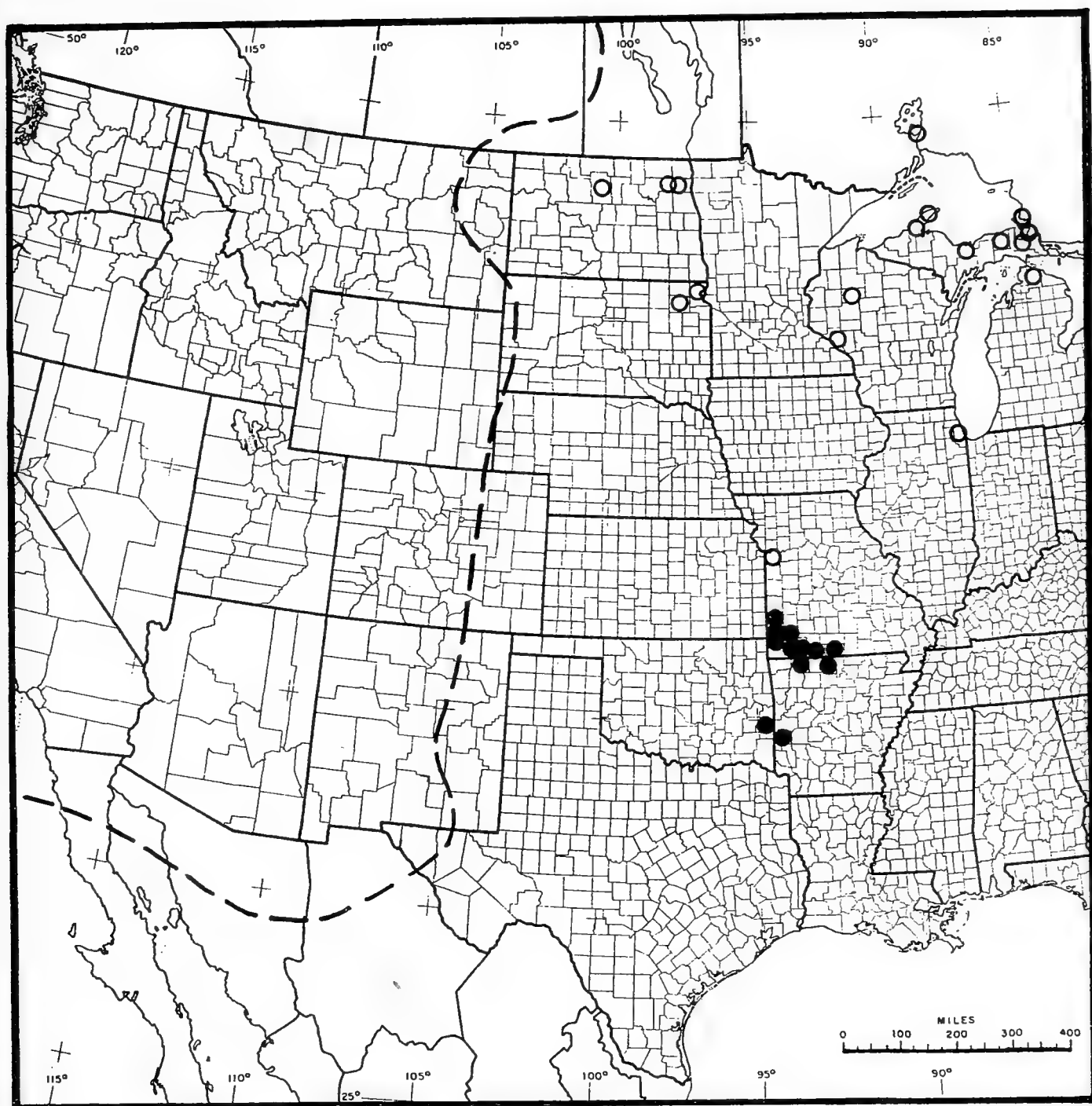


Figure 2b. *Elymus glaucus*: mapped county records of ssp. *mackenzii* and outlying eastern records referable to ssp. *glaucus* (central North America). Solid dots show ssp. *mackenzii* (see text for collection details); open dots show other eastern records of *E. glaucus*, including plants that are similar to *E. trachycaulus*, based on authorities (e.g., Bowden 1954; Gould 1975; McGregor et al. 1986; Voss 1972), and on scattered collections seen by the author. The dashed line shows approximate eastern boundary of main range of *E. glaucus* based on reliable data accumulated by M. E. Barkworth (pers. comm.); there are no other records between this line and ssp. *mackenzii*.

←

Figure 2a. *Elymus glaucus* ssp. *mackenzii* (Bush) J.J.N. Campbell.—A. Habit.—B. Upper portion of culm with mature spike, viewed on plane with alternating spread of spikelets.—C1. Sheath summit and blade base.—C2. Adaxial blade pubescence.—D1. Mature rachis internode and glumes, viewed in plane of spikelet spread (with abaxial view of central glume of spikelet, and largely side view of lateral glume); note lack of prompt disarticulation by rachilla base from rachis.—D2. Spikelet, with lateral view of florets.—E. Mature floret in abaxial view (left) and adaxial view (right); note narrowly truncate palea summit almost equalling lemma body.—F. Cross-section of mature glume base, showing surficial vascular bundles on abaxial side.—G. Cross-section of central rachis internode, showing lack of angles on abaxial side. Drawn from robust material of H.H. Iltis et al. 1 (WIS, KY).

- 2. Blades usually 4–13 mm wide, glabrous to strigose above, or occasionally pilose to hirsute with hairs of fairly uniform length; glume awns 1–5 mm.
- 3. Blades strigose, pilose, or hirsute; lemmas awns to 20 mm. ssp. *jepsonii* (Burt Davy) Gould
- 3. Blades glabrous, scabrous, or sparsely strigose; lemma awns to 35 mm ssp. *glaucus*
- 2. Blades usually 3–8 mm wide, densely short-pilose and with scattered longer hairs above; glume awns 3–8 mm ssp. *mackenzii* (Bush) J.J.N. Campbell

Very few vascular plants are known to have such striking disjunctions from western ranges to the Ozarks or Ouachitas, as is the case in *Elymus glaucus*. *Mimulus floribundus* Douglas ex Lindl. (Scrophulariaceae) is a widespread species of the Rocky Mountains that occurs on wet dripping cliffs in the Ozark Mountains of Arkansas (Moore 1959; Smith 1991). Like *E. glaucus*, the eastern plants of *M. floribundus* appear to have some morphological distinction, but in this case a subspecific epithet has not yet been published (H.H. Iltis, pers. comm., April 1999).

ACKNOWLEDGMENTS

For their assistance and encouragement, I am particularly grateful to Mary Barkworth, Ralph Brooks, Hugh Iltis, Dan Nicholson, Max Medley, John and Charlotte Reeder, Rob Soreng, and George Yatskievych. I thank the curators of the various herbaria where I have studied material or obtained loans.

LITERATURE CITED

Bowden, W. M. 1964. Cytotaxonomy of the species and interspecific hybrids of the genus *Elymus* in Canada and neighboring areas. *Canad. J. Bot.* 42:547–601.

Brooks, R. E. 1974. Intraspecific variation in *Elymus virginicus* (Gramineae) in the central United States. M.A. Thesis. University of Kansas, Lawrence, KS.

Bush, B. F. 1926. The Missouri species of *Elymus*. *Am. Midl. Naturalist* 10:49–88.

Campbell, J. J. N., M. Evans, M. E. Medley, and N. L. Taylor. 1988. Buffalo clovers in Kentucky (*Trifolium stoloniferum* and *T. reflexum*): historical records, presettlement environment, rediscovery, endangered status, cultivation and chromosome number. *Rhodora* 90:399–418.

Campbell, J. J. N. 1995. New combinations in eastern North American *Elymus* (Poaceae). *Novon* 5:128.

Campbell, J. J. N. 1996. Proposal to conserve *Elymus vir-*

ginicus (Poaceae) with a conserved type. *Taxon* 45:128–129.

Campbell, J. J. N., J. R. Abbott, R. R. Cicerello, J. D. Kiser, J. R. MacGregor, and J. G. Palis. 1994. Cooperative inventory of endangered, threatened, sensitive and rare species, Daniel Boone National Forest, London Ranger District. Kentucky State Nature Preserves Commission, Frankfort, KY.

Dewey, L. H. 1892. [Gramineae.] In J. H. Coulter (ed). *Manual of the phanerogams and pteridophytes of western Texas*. *Contrib. U.S. Natl. Herb.* 2(3):347–588.

Dore, W. G., and J. McNeill. 1980. *Grasses of Ontario*. Biosystematics Research Institute, Ottawa, Ontario. Research Branch, Agriculture Canada, Monograph 26.

Gould, F. W. 1975. *The grasses of Texas*. Texas A&M University Press, College Station, TX.

Hickman, J. C. (ed). 1993. *The Jepson manual of higher plants of California*. University of California Press, Berkeley, CA.

Holmgren, P. K., N. H. Holmgren, and L. C. Barnett. 1990. *Index herbariorum. Part I: The herbaria of the world*. International Association for Plant Taxonomy, New York Botanical Garden, Bronx, NY.

Jones, M. E. 1912. [*Elymus minor* comb. nov.] *Contrib. Western Bot.* 14:20.

Little, E. L. 1971. *Atlas of United States trees. Vol. 1. Conifers and important hardwoods*. USDA Misc. Publ. 1146.

McGregor, R. L., T. M. Barkley, R. E. Brooks, and E. K. Schofield. 1986. *Flora of the Great Plains*. Great Plains Flora Association, University Press of Kansas, Lawrence, KS.

Moore, D. M. 1954. An ecological and botanical survey: Table Rock Reservoir. Technical Report to National Park Service, University of Arkansas, Fayetteville, AR.

Moore, D. M. 1959. *Mimulus floribundus* in Arkansas. *Southwest. Naturalist* 3:217–219.

Smith, E. B. 1991. *An atlas and annotated list of the vascular plants of Arkansas*, 2nd ed. University of Arkansas, Fayetteville, AR.

Snyder, L. A. 1950. Morphological variability and hybrid development in *Elymus glaucus*. *Am. J. Bot.* 37:628–635.

Snyder, L. A. 1951. Cytology of inter-strain hybrids and the probable origin of variability in *Elymus glaucus*. *Am. J. Bot.* 38:195–202.

Stebbins, G. L., and L. A. Snyder. 1956. Artificial and natural hybrids in the Gramineae, Tribe Hordeae. IX. Hybrids between western and eastern North American species. *Am. J. Bot.* 43:305–312.

Stebbins, G. L. 1957. The hybrid origin of microspecies in the *Elymus glaucus* complex. *Proc. Internat. Genetics Symposia*, 1956:336–340.

Steyermark, J. A. 1963. *Flora of Missouri*. Iowa State University Press, Ames, IA.

Voss, E. G. 1972. *Michigan flora. Part I. Gymnosperms and monocots*. Cranbrook Institute of Science, Bloomfield Hills, MI; University of Michigan, Ann Arbor, MI.

Yatskievych, G. 1999. Steyermark's flora of Missouri, Vol. 1. Missouri Botanical Garden Press, St. Louis, MO.

APPENDIX: COUNTIES AND
HERBARIUM SOURCES FOR
SPECIMENS OF *ELYMUS*
MACGREGORII ANNOTATED BY THE
AUTHOR.

I have annotated collections as *Elymus macgregorii* (or *E. virginicus* var. *minor*) from the following counties. Herbarium acronyms (Holmgren et al. 1990) are in parentheses. Uncertain identifications due to incomplete or depauperate specimens or probable hybrids are generally excluded unless they represent possible county records (with caveats noted in parentheses). However, a frequent, mostly southwestern, variant that may be transitional to *E. virginicus* var. *jejunus* is included and shown by asterisks.

CANADA. NOVA SCOTIA: "Bass River" (MADI, US)? [30 Jul 1875, *J. Fowler s.n.*—this northernmost specimen is atypical and might be at least transitional to *E. virginicus*, but is included here as the only possible record seen so far from Canada].

U.S.A. ALABAMA: Bibb (VDB); St. Clair (NCU). ARKANSAS: Ashley (UARK*); Baxter (UARK*); Benton (MADI, NCU,* UARK*); Boone (KANU*); Bradley (VDB); Fulton (UARK*); Hot Spring (US); Independence (KANU); Montgomery (VDB); Newton (BEREA*, UARK*); Polk (ISC, VDB); Prairie (ISC); Washington (NCU,* UARK,* US*). CONNECTICUT: New Haven (US). DISTRICT OF COLUMBIA (ISC, MADI). FLORIDA: Leon (TEX) [*Godfrey 84158*, "common in ditches"—could this be adventive?]. GEORGIA: Clarke (US*). ILLINOIS: Fulton (US); Jersey (KNK); Knox (MO); Peoria (US); Union (MADI, KNK). INDIANA: Parke (US); Putnam (US); Wayne (US). IOWA: Boone (ISC); Clay (ISC, MO*); Dickinson (ISC); Emmet (ISC); Jefferson (ISC*); Lee (ISC); Lyon (ISC); Mahaska (ISC); Story (MO,* NCU*); Winneshiek (ISC*). KANSAS: Allen (KANU); Anderson (KANU); Atchison (KANU*); Brown (KANU); Butler (KANU); Cherokee (KANU); Cowley (KANU, MADI); Dickinson (KANU); Doniphan (KANU*); Douglas (KANU); Franklin (KANU); Jackson (KANU); Jefferson (KANU); Kingman (KANU); Leavenworth (KANU); Miami

(KANU); Morris (KANU); Nemaha (KANU); Shawnee (KANU); Wabaunsee (KANU); Wyandotte (KANU). KENTUCKY: Anderson (EKY); Barren (KY); Boone (KNK); Calloway (NCU); Campbell (KNK, NCU); Casey (BEREA, KY); Christian (VDB); Clark (KY); Clay (MO); Fayette (KY, US); Franklin (KY); Fulton (KY, VDB); Grant (KNK); Graves (VDB); Green (EKY); Hickman (KY); Jefferson (KY, NCU, TEX, UTC); Jessamine (KY); Kenton (KNK, VDB); Laurel (BEREA); Livingston (KY); McCracken (KY); Madison (KY); Oldham (KY); Owsley (KY); Pendleton (NCU); Pulaski (KY); Rowan (KY); Spencer (KY); Trimble (KY); Warren (WKY); Wayne (MORE); Wolfe (KY); Woodford (KY). LOUISIANA: Bienville (US); De Soto (US); Jefferson? (VDB—perhaps transitional to *E. glaberrimus*); Ouachita (NCU); St. Tammany (ISC). MAINE: Knox (NCU); Oxford (GH); Penobscot (ISC); Piscataquis (US); Washington (NCU); York (NCU, US). MARYLAND: Garrett (US); Montgomery (TEX); Washington (KANU). MASSACHUSETTS: Essex (US); Hampshire (MADI); Worcester (MADI, TEX). MISSISSIPPI: Forrest (US); Tunica (NCU). MISSOURI: Barry (MADI*); Boone (MO); Christian (ISC*); Cole (MO); Dade (MO, US); Franklin (MO); Greene (NCU); Jackson (ARIZ, ISC, MO, US); Jasper (MO, US); Jefferson (KANU, MO, NCU, UTC*); Lafayette (MO); McDonald (MO); Monroe (MO); Morgan (MO); Oregon (MO); Pettis (MO); Phelps (MO); Pike (MO); Platte (MO); Ralls (MO, US, UTC); St. Charles (MO, UTC); St. Clair (TENN); Ste. Genevieve (MO); St. Louis (ISC, MO); Shannon (MO); Taney (MO); Texas (MO); Warren (MO). NEBRASKA: Richardson (KANU, US*). NEW HAMPSHIRE: Cheshire (GH); Coos (ISC, TEX, US); Hillsborough (GH, MADI, MO, TENN); Strafford (MADI). NEW YORK: Cattaraugus (MO); Tompkins (MO). NORTH CAROLINA: Alamance (US); Chatham (NCU*); Harnett (NCU); Haywood (NCU); Henderson (NCU); Jones (NCU); Lee (NCU); Martin (NCU); Northampton (NCU); Polk (NCU*); Stokes (NCU); Wilson (NCU). NORTH DAKOTA: McClean? (KANU—at least transitional to *E. virginicus*). OHIO: Ashland (TENN); Franklin (VDB); Hamilton (US); Huron (NCU). OKLAHOMA: Comanche (US); Kay (KANU); Muskogee (US); Rog-

ers (VDB*). PENNSYLVANIA: Elk (ISC); Luzerne (TEX); Snyder (TEX); Warren (ISC). RHODE ISLAND: Providence (GH). SOUTH DAKOTA: Clay? (MO—perhaps transitional to *E. virginicus*); Roberts (ISC). TENNESSEE: Bledsoe (VDB); Davidson (KANU, MADI, US, VDB); Dickson (VDB); Smith (VDB); Williamson (VDB). TEXAS: Austin (MO,* TEX, US); Bexar (MADI,* MO,* TEX); Blanco (KANU, TEX); Brazoria (TEX); Brazos (TEX*); Brown (TEX*); Burleson (US*); Collin (MO*); Comal (MO, TEX,* US*); Dallas (MO*); Dimmit (US*); Fort Bend (TEX*); Galveston (TEX*); Gillespie (TEX*); Gonzales (TEX); Harris (TEX*);

Hays (TEX); Jim Wells (US*); Llano (TEX); Medina (TEX); Nacogdoches (US); Nueches (MO); Refugio (TEX); San Patricio (ISC, MO, TEX); San Saba (MO); Tarrant (TEX,* US*); Taylor (ISC,* MO, TEX); Travis (ISC*, MO,* TEX); Val Verde (TEX*); Wichita (TEX*). VERMONT: Essex (US). VIRGINIA: Buckingham (NCU); Clarke (NCU); Loudoun (NCU); Montgomery (NCU*); Powhatan (NCU). WEST VIRGINIA: Jefferson (NCU); Morgan (NCU); Tucker (TEX); Upshur (NCU). WISCONSIN: Dodge (MADI); Fond Du Lac (MADI); Grant (MADI); Iowa (MADI); Outagamie (MADI); Vernon (MADI).

***Proterometra macrostoma* (Digenea: Azygiidae): Distome Emergence From the Cercarial Tail and Subsequent Development in the Definitive Host**

Ronald Rosen, Kelly Adams, Emilia Boiadgieva, and Jessica Schuster

Department of Biology, Berea College, Berea, Kentucky 40404

ABSTRACT

The objectives of our study were (1) to evaluate effects of pH and pepsin on emergence of the *Proterometra macrostoma* distome body from its cercarial tail and (2) to examine morphological changes associated with maturation of this worm in the sunfish definitive host. Distome emergence from the cercarial tail was significantly faster at low pH (1.5–2.5). Addition of 0.5% pepsin appeared to accelerate this process. The number and maximum size of eggs increased in adult worms over the initial 18 and 24 days, respectively, in experimentally infected bluegill, *Lepomis macrochirus*. No other trends for change in worm size were noted. Mature eggs containing miracidia were first observed by day 18 postinfection.

INTRODUCTION

Proterometra macrostoma is a digenetic trematode widely distributed in eastern United States. The complete life cycle was first described by Horsfall (1933, 1934). The adult worm is found in the esophagus and stomach of sunfishes (Family Centrarchidae). Eggs containing fully developed miracidia are released into water with fecal material and are subsequently ingested by snails in the genus *Elimia*. Intramolluscan stages include sporocysts, rediae, and cercariae. During emergence from the snail, the distome body of the cercaria enters a vesicle/cavity within the cercarial tail and detaches from the latter structure. After it is released into the water, the cercaria's swimming behavior and large size—tailstem 4–8.9 mm long (LaBeau and Peters 1995)—make it attractive to potential definitive hosts, which rapidly ingest the worm. The progenetic (i.e., containing in uterus eggs in early cleavage) distome body is then liberated from the cercarial tail in the fish stomach and matures directly into an adult worm in the host stomach or esophagus.

The time frame and conditions that promote the emergence of the distome body from the cercarial tail have not been quantified. Horsfall (1934) reported emergence of *P. macrostoma* from the cercarial tail in a 1.0% solution of HCl, but it was unclear what pH, temperature, and time frame were used. Similarly, the subsequent development of this “liberated” distome in the definitive host has been based on morphological comparisons between

the distome body within the cercaria vs. the adult worm (Horsfall 1934). The time frame for changes associated with maturation of the adult worm in sunfishes has not been previously described.

The objectives of our study were (1) to evaluate the effect of pH and pepsin on emergence of the distome body from the cercarial tail of *P. macrostoma* and (2) to establish a time line for possible morphological changes during maturation in sunfish definitive hosts.

METHODS

General Methods

Snails were collected in June and July 1998 from North Elkhorn Creek (38° 11' 00" N, 84° 29' 19" W) in Scott County, Kentucky. Snails were maintained under a protocol similar to that described by Riley and Uglem (1995). They were placed in white enamel pans filled with filtered creek water, held at 20–25°C under continuous light, and fed lettuce ad libitum. The water in the pans was changed every 2 days. When cercariae were required for experiments, any previously emerged worms were removed from these pans. The snail cultures were then placed in an environmental chamber in the dark at 20°C, which promoted a copious release of new cercariae within 2 hours.

Distome Emergence

To assess the effect of pH and pepsin on distome emergence from the cercarial tail, a Ringer's solution for cold-blooded vertebrates

(i.e., 6.5 g/liter NaCl, 0.05 g/liter KCl, 0.16 g/liter $\text{CaCl}_2 \times 2\text{H}_2\text{O}$, 0.39 g/liter $\text{MgSO}_4 \times 7\text{H}_2\text{O}$, and 0.2 g/liter NaHCO_3) was acidified with concentrated HCl to obtain pHs of 1.5, 2.0, 2.5, 3.0, 3.5, and 4.0. These values were within the physiological range (i.e., pH 1–5) of bluegill gastric juice reported by Norris et al. (1973). For the pH + pepsin experiments, 0.5% pepsin (Sigma: 1:2500; activity 600–1800 units per mg protein) solutions were made with each of the acidified salines. Cercariae less than 2.5 hours postemergence were used in these experiments. They were pipetted into individual beakers containing a particular pre-cooled (20°C) acidified saline or acidified saline + pepsin, incubated at 20°C, and checked for complete distome emergence from the cercarial tail every 5 minutes for 1 hour. Each experiment consisted of six treatments and 60 worms, with 10 cercariae/treatment (=pH). Both the pH and pH + pepsin experiments were repeated three times.

A mean \pm SE (% distome emergence) was calculated for each time period and pH based on the three replicate treatments. A chi-square goodness-of-fit test was used to determine if significant differences in the number of emerging distomes existed due to pH at 5 and 30 minutes for both the acidified saline and acidified saline + pepsin experiments. A 2×2 chi-square contingency test was used to assess differences in numbers of digeneans emerging from cercarial tails in acidified saline vs. acidified saline + pepsin at 5 and 30 minutes for each pH level assessed. A probability of $P < 0.05$ was considered significant for all statistical tests.

Fish Infections

Young, hatchery-reared (Ken Jacobs, Bowling Green, KY) bluegill, *Lepomis macrochirus* (mean fork length = 5.2 cm), were used in this experiment. Fish were maintained in aerated 10 and 15 liter aquariums at 24.6°C and fed TetraMin (Tetra Sales, Blacksburg, VA). Prior to infection, one bluegill was placed into a 3.3 liter tank. After a 1–3 minute acclimation period, two cercariae were released into this tank. Visual confirmation of cercariae ingestion was made, and then the exposed fish was placed into a new aquarium. In addition, several bluegill were exposed to cercariae in mass

and subsequently maintained in the manner described above.

Every 3 days, beginning at day 0 (at 5 and 15 minute postinfection [PI]) and ending on day 24 PI, 3–5 fish were sacrificed and necropsied. Worms were removed from the stomach, placed on a slide under a coverslip, and examined with a compound microscope. The number and developmental stage of eggs were recorded. Developmental stages were based on the descriptions of Horsfall (1934): (1) Stage I—eggs containing a clear mass at their anterior end and a large vitelline mass at the posterior end, (2) Stage II—the vitelline mass in eggs is less apparent and signs of advanced cleavage are more obvious, and (3) Stage III—further reduction in the vitelline mass, darkening of the egg into a yellow color, and appearance of bristle plates associated with the developing miracidium at the opercular end. The worm was then fixed and flattened under a coverslip with FAA (formalin-alcohol-acetic acid). Flukes were stained with Semichon's carmine, and permanent slides were made. Length and width measurements of worm size, oral sucker, acetabulum, pharynx, testes, and eggs were obtained from fixed/stained specimens with an ocular micrometer. Means \pm SE in micrometers were calculated from these measurements for five time-intervals.

RESULTS

The number of emergent distomes in the six acidified salines without 0.5% pepsin (Figure 1) was significantly different at 5 minutes ($\chi^2 = 24.5714$; $df = 5$) and 30 minutes ($\chi^2 = 42.4286$; $df = 5$). Similarly, the number of emerging digeneans in the six acidified salines with 0.5% pepsin (Figure 1) was significantly different at 5 minutes ($\chi^2 = 53.3019$; $df = 5$) and 30 minutes ($\chi^2 = 65.9895$; $df = 5$). The overall trend suggested that emergence of *P. macrostoma* from the cercarial tail was facilitated at lower pH levels (i.e., 1.5–2.5; Figure 1).

Significant differences were noted in emergence of digeneans from the cercarial tail when the effect of acidified salines was compared to acidified salines + 0.5% pepsin at 5 minutes (pH 1.5— $\chi^2 = 20.3175$, $df = 1$; pH 2.0— $\chi^2 = 13.6111$, $df = 1$; pH 2.5— $\chi^2 = 5.5430$, $df = 1$) and 30 minutes (pH 1.5— $\chi^2 = 15.5556$, $df = 1$; pH 2.0— $\chi^2 = 12.0000$, df

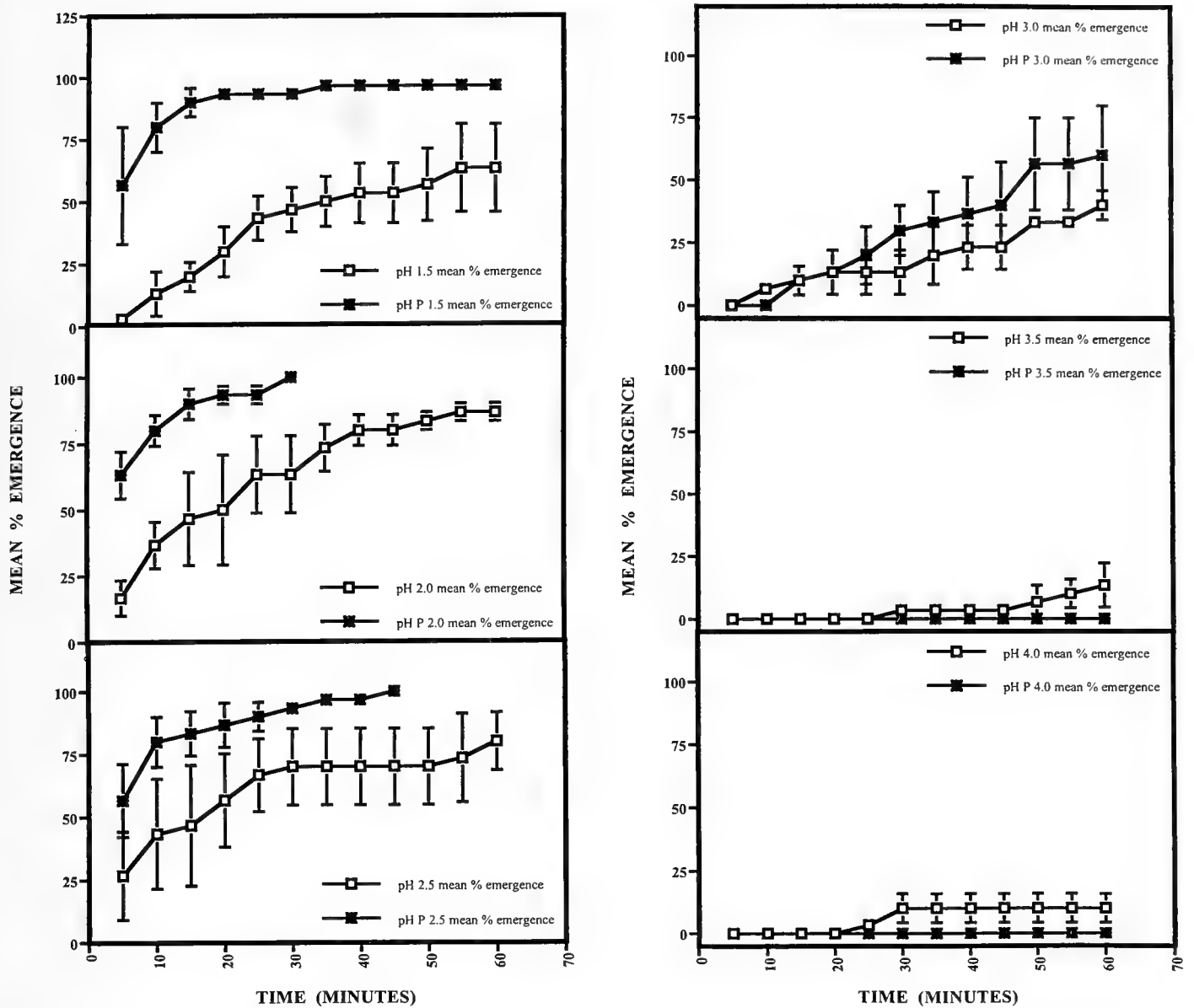


Figure 1. Effect of pH and pepsin on mean % distome emergence from the tail of *Proterometra macrostoma* over 60 minutes at 20°C. Means represent the average % emergence \pm standard error (SE) from the three replicates with 10 cercariae/replicate. (pH = acidified saline only; pH P = acidified saline + 0.5% pepsin)

= 1; pH 2.5— $\chi^2 = 4.320$, $df = 1$). Emergence of *P. macrostoma* appeared to be greatly enhanced by the addition of pepsin to the most acidic pH's (i.e., 1.5–2.5). No significant differences were noted between these two treatments at the less acidic pH's (i.e., 3.0, 3.5, and 4.0), and overall emergence was minimal (Figure 1).

Within 15 minutes PI, all distomes had emerged from their cercarial tails in experimental infections of bluegill. No obvious trends for subsequent size increases were noted for worms in such infections other than changes in maximum egg size (Figure 2). The mean number of eggs/worm also showed a gradual increase from day 0 (13.8) to days 14–

18 (120.7) PI, followed by a slight decline in number between days 21–24 PI (Figure 3).

Changes in the general morphology/development of eggs were noted over the course of the experiment. Stage I eggs were the only eggs observed in adult worms from days 0 to 9 PI. After day 9 PI, a transformation into Stage II eggs was observed. It should be noted that Stage I egg production continued during this second stage, and thus two “populations” of eggs were present in worms at this time. Stage III (mature) eggs were apparent by day 18 PI. All three egg stages were observed in worms by this time; mature eggs were dispersed at the anterior end of the adult worm; immature eggs, at the posterior end.

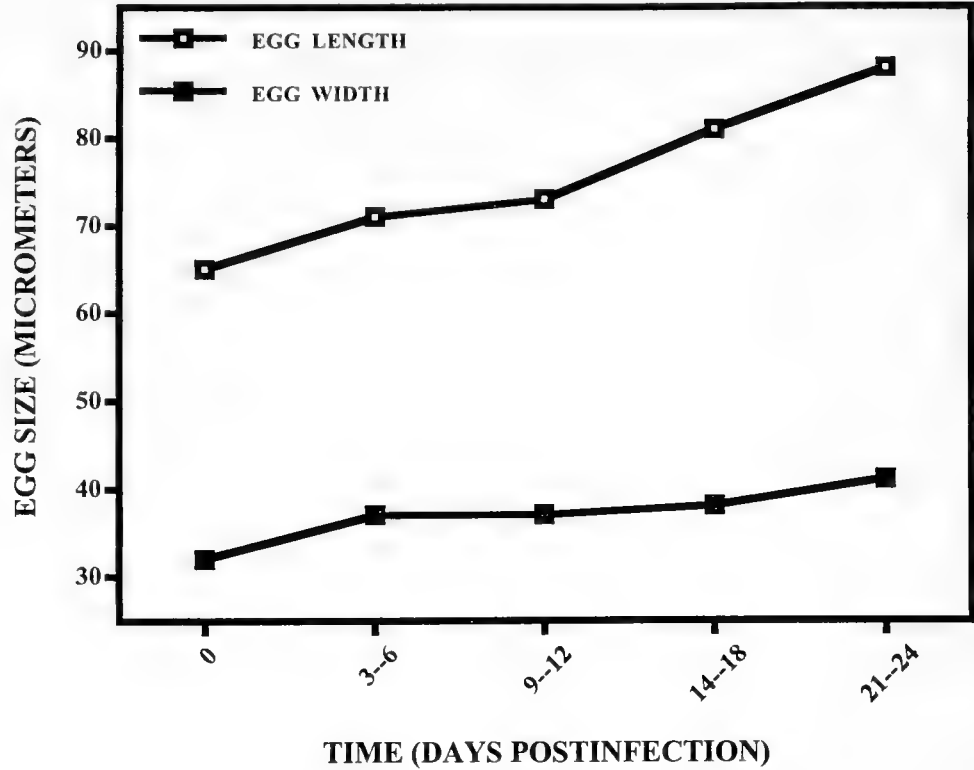


Figure 2. Mean lengths and widths of largest eggs in developing distomes of *Proterometra macrostoma* from experimental infections of bluegill, *Lepomis macrochirus*, over 24 days postinfection at 24.6°C. (number of eggs measured: day 0, n = 17; days 3–6, n = 42; days 9–12, n = 30; days 14–18, n = 33; days 21–24, n = 30)

DISCUSSION

Trematodes in the family Azygiidae are unusual in that they bypass a metacercarial stage, which is normally encysted in host tissue. Worms in this family are “encysted” in the tail of their own cercaria; thus there is no host tissue to be digested. Only triggers in the stomach (e.g., pH and pepsin) seem essential for emergence and subsequent establishment of the distome.

The high percentage of distome emergence in more acidic salines (pH 1.5–2.5) with 0.5% pepsin at 5 minutes and at 30 minutes (Figure 1) was similar to the rapid emergence observed in the experimental infections of bluegill at day 0 PI. In vivo, some *P. macrostoma* were recovered as emerged distomes at 5 minutes PI, and all had emerged from cercarial tails by 15 minutes PI. The similarity in rate of emergence in vivo vs. in vitro suggests that a pH range of 1.5–2.5 with a pepsin concentration of 0.5% approximates the conditions inducing emergence of the worm in the fish stomach. This is further corroborated by the work of Norris et al. (1973), who examined the rates of gastric acid and pepsin secretion into the gastric juice of bluegills. They found that bluegill gastric juice decreased from a pH of

5 to a pH of 1–2 immediately following ingestion of a simulated meal. Pepsin activity would be most pronounced in such a strongly acidic environment, further enhancing distome release from the cercarial tail.

Cercariae of *P. dickermani* (Anderson and Anderson 1963) and *P. autraini* (LaBeau and Peters 1995) contain eggs with fully developed miracidia, while only undeveloped eggs have been reported in cercariae from other species in this genus (Anderson and Anderson 1967; Horsfall 1934; Smith 1936). It has been suggested that a signal from the fish definitive host triggers maturation in some of these latter species of *Proterometra* (Braham et al. 1996). This “signal” must be focused on increased egg production as well as on egg maturation; other structures within these distomes appear to be fully developed following their emergence from the cercarial tail.

Dickerman (1934) observed that the only differences between the distome of the *P. macrostoma* cercaria and that of the adult worm were size and number of eggs present in utero. Similarly, Horsfall (1934) noted that, “the numerous eggs of the adult (*P. macrostoma*) mask certain structures which are conspicuous in the larval distome; otherwise the

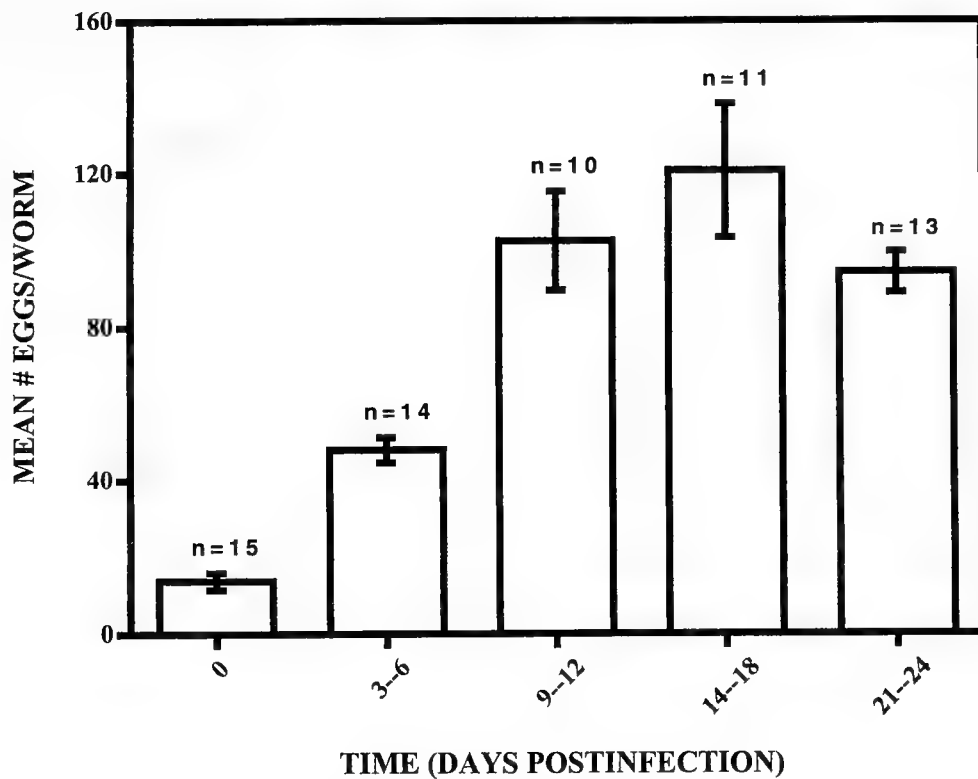


Figure 3. Mean \pm SE eggs/*Proterometra macrostoma* distomes obtained from experimental infections of bluegill, *Lepomis macrochirus*, over 24 days postinfection at 24.6°C. (n = number of worms assessed at each time interval)

general appearance (of the worm) is the same.” Our observation of increased egg production in *P. macrostoma* over the initial 2.5 weeks of infection in bluegill corroborates these observations. Increased egg production also occurs in *P. albacauda* (Anderson and Anderson 1967), *P. catenaria* (Anderson and Anderson 1967), *P. edneyi* (Uglen and Aliff 1984), *P. sagittaria* (Dickerman 1946), and *P. septimae* (Anderson and Anderson 1967) following release from their cercarial tails in the definitive host.

Egg production appears to be continuous once the distome of *P. macrostoma* is released in the fish stomach as evidenced by (1) the steady increase in egg number over time (up to day 18 PI) and (2) the presence of Type I eggs (i.e., early cleavage) in older fish infections. Decrease in egg number between days 21–24 PI may coincide with initiation of egg release from the adult worm into the host digestive tract, but further work will be required to verify this.

In our study, *P. macrostoma* eggs required 18–24 days at 24.6°C to develop miracidia. Horsfall (1934) indicated that 15–30 days are required depending on the number of eggs contained within the digenean prior to its emergence from the cercarial tail. In addition

to the progenetic state of these distomes, proposed *P. macrostoma* strain differences (Dickerman 1945; Riley and Uglen 1995) may affect maturation time.

Current work is being conducted in our lab using molecular techniques to determine the validity of these *P. macrostoma* strains.

ACKNOWLEDGMENTS

Our study was supported by grants from the Andrew Mellon Foundation (Appalachian College Association) and the Undergraduate Research Creative Projects Program (URCPP) at Berea College. We also acknowledge the Hollingsworth family for permitting collection of snails on their property.

LITERATURE CITED

- Anderson, M. G., and F. M. Anderson. 1963. Life history of *Proterometra dickermani* Anderson, 1962. J. Parasitol. 49:275–280.
- Anderson, M. G., and F. M. Anderson. 1967. The life histories of *Proterometra albacauda* and *Proterometra septimae*, spp. n. (Trematoda: Azygiidae) and a redescription of *Proterometra catenaria* Smith, 1934. J. Parasitol. 53:31–37.
- Braham, G. L., M. Riley, and G. L. Uglen. 1996. Infectivity and the cercarial tail chamber in *Proterometra macrostoma*. J. Helminthol. 70:169–170.
- Dickerman, E. E. 1934. Studies on the trematode family

- Azygiidae. I. The morphology and life cycle of *Proterometra macrostoma* Horsfall. Trans. Am. Microscop. Soc. 53:8–21.
- Dickerman, E. E. 1945. Studies on the trematode family Azygiidae. II. Parthenitae and cercariae of *Proterometra macrostoma* (Faust). Trans. Am. Microscop. Soc. 64: 138–144.
- Dickerman, E. E. 1946. Studies on the trematode family Azygiidae. III. The morphology and life cycle of *Proterometra sagittaria* n. sp. Trans. Am. Microscop. Soc. 65:37–44.
- Horsfall, M. W. 1933. Development of *Cercaria macrostoma* Faust into *Proterometra* (nov. gen.) *macrostoma*. Science 78:175–176.
- Horsfall, M. W. 1934. Studies on the life history and morphology of the cystocercous cercariae. Trans. Am. Microscop. Soc. 53:311–347.
- LaBeau, M. R., and L. E. Peters. 1995. *Proterometra atraini* n. sp. (Digenea: Azygiidae) from Michigan's upper peninsula and a key to species of *Proterometra*. J. Parasitol. 81:442–445.
- Norris, J. S., D. O. Norris, and J. T. Windell. 1973. Effect of simulated meal size on gastric acid and pepsin secretory rates in bluegill (*Lepomis macrochirus*). J. Fish. Res. Board Canada. 30:201–204.
- Riley, M. W., and G. L. Uglem. 1995. *Proterometra macrostoma* (Digenea: Azygiidae): variations in cercarial morphology and physiology. Parasitology 110:429–436.
- Smith, S. 1936. Life cycle studies of *Cercaria hodgesiana* and *Cercaria melanophora*. J. Alabama Acad. Sci. 8:30–32.
- Uglem, G. L., and J. V. Aliff. 1984. *Proterometra edneyi* n. sp. (Digenea: Azygiidae): behavior and distribution of acetylcholinesterase in cercariae. Trans. Am. Microscop. Soc. 103:383–391.

New State Records and New Available Names for Species of Kentucky Moths (Insecta: Lepidoptera)

Charles V. Covell Jr.

Department of Biology, University of Louisville, Louisville, Kentucky 40292-0001

Loran D. Gibson

2727 Running Creek Drive, Florence, Kentucky 41042

and

Donald J. Wright

3349 Morrison Avenue, Cincinnati, Ohio 45220-1430

ABSTRACT

The authors add records of 35 moth species to the list of Lepidoptera known in Kentucky, bringing the total to 2423 from the 2388 published in the Covell (1999) annotated checklist. These are in the families Gracillariidae (1), Oecophoridae (1), Gelechiidae (3), Tortricidae (4), Crambidae (16), Pterophoridae (2), Geometridae (1) and Noctuidae (7). In addition scientific names are given for the three “*Chionodes* undescribed species” (Gelechiidae) listed as such by Covell (1999) and recently made available by Hodges (1999).

INTRODUCTION

Intensive collecting, identification, and monitoring of the moth and butterfly fauna of Kentucky culminated in December 1999 with the publication of Covell (1999), in which 2388 species of moths and butterflies were documented. Even at that time there were additional species to add. And since that publication came out names have been made available for three species that were indicated under generic headings as “species” because we could not cite unpublished names. Thirty-eight species are added to the state list in this paper, bringing the total to 2426. This is the first supplement to Covell (1999).

We thank the following specialists for identifying specimens from which most of these records resulted: Bernard Landry, William E. Miller, Herb Neunzig, Eric L. Quinter, Michael Sabourin, David Wagner, and Reed Watkins.

Below are listed the new additions and names in order of the numbers assigned in the Hodges et al. (1983) checklist. Specimens on which these identifications are based are in the Lovell Insect Museum at the University of Louisville and in the private collections of those cited as the collectors. Those cited from Hodges (1999) are located in the National

Museum of Natural History, Washington, D.C., and the Philadelphia Academy of Natural Sciences collection.

GRACILLARIIDAE

- 0606 *Caloptilia fraxinella* (Ely)
Boone Co., Big Bone Lick State Park, larva collected on *Fraxinus* 4 Jun 1995; adult emerged 5 Jun 1995; D.J. Wright.

OECOPHORIDAE

- 1015 *Antaeotricha osseola* (Wlsm.)
Laurel Co., junction of forest service roads 121 and 4158, larva collected on *Quercus montana*, 18 May 1996, adult emerged 31 Jul 1996; Rowan Co., east end of Clack Mountain Road West, 26 Aug 1994; both collected by D.J. Wright.

GELECHIIDAE

- 2061.1 *Chionodes hapsus* Hodges
A paratype female of this species was listed by Hodges (1999, p. 56) from Fleming Co., Fleming, 31 May 1938, A.F. Braun. This is not one of the “*C. undescribed species*” listed in Covell (1999, p. 37).
- 2061.3 *Chionodes suasor* Hodges
A paratype male of this species was listed by Hodges (1999, p. 58) from Rowan Co., Morehead, 30 Jun 1960, collected by Lewis and Freeman. This is not one of the “*C. undescribed species*” listed in Covell (1999, p. 37).

2119.2 *Chionodes sevir* Hodges
This name validates the entry under this checklist number in Covell (1999, p. 37). A paratype male is listed in Hodges (1999, p. 138) with the following data: Fulton Co., Fulton, 19 Sep 1975, C. C. Cornett. See Covell (1999, p. 37).

2119.1 *Chionodes baro* Hodges
A paratype male is listed by Hodges (1999, p. 145) from Bell Co., Pine Mountain State Park, 18 Jul 1975, A.J. Brownell. This is the name that validates the entry under this checklist number in Covell (1999, p. 37). The designation "Harlan Co." in that entry was an error.

2119.3 *Chionodes adamas* Hodges
This is the name that validates the entry under this checklist number in Covell (1999, p. 37). Paratype data as published by Hodges (1999, p. 151) follow: "Clack Mountain, Rowan County; 19, 22 June 1941; A.F. Braun (2 females). Otter Creek Park, Meade County, 8 October, 1979, C.V. Covell Jr. (1 female). Same locality, 17 March, 1987, B.S. Nichols (1 male). Rd 9B, Indian East Fork, Kelley Br., 720', Manifee [sic] County; 9–18 August, 1985; J.S. Nordin (2 males, 1 female). Tunnel Ridge Road, Powell County; 4–11 March, 1989; D.J. Wright (1 male, 2 females)."

2066.1 *Chionodes aruns* Hodges
Hodges (1999, p. 189) described this species from Texas, and although he lists Kentucky specimens of the species, they were not included in the paratype series. Their data are quoted as follows: "Cumberland National Forest, Pulaski County, Kentucky; 26 April 1939; A.F. Braun (1 female). Lick Fork, Rowan County, Kentucky; 4 April 1938; A.F. Braun (1 male). Cascade Caves, Carter County, Kentucky; 5 May 1956; A.F. Braun (1 female)."

TORTRICIDAE

2717 *Endopiza yaracana* (Kft.)
Powell Co., Red River Gorge, Tunnel Ridge Road; 3 May 1991, D.J. Wright.

2795 *Olethreutes tiliana* (Heinr.)
Boone Co., Middle Creek Road, larvae collected on *Tilia neglecta* 5 May 1993, two females emerged on 30 May 1993; Laurel Co., Rockcastle Campground, larva collected on *Tilia* 1 May 1993, adults emerged 27 May and 3 and 10 Jun 1993, D.J. Wright; Laurel Co., Daniel Boone National Forest, Rockcastle Recreation Area, larva on *Tilia neglecta* collected 1 May 1993, two females emerged 25 and 28 May 1993.

2885 *Rhyacionia aktita* W.E. Miller
Laurel Co., Daniel Boone National Forest, Forest Service Road 131, 2 miles from State Road 3497, 19 Apr 1992, D.J. Wright; powerline corridor east side of south end of Forest Service Road 775, 11

Apr 1997, L.D. Gibson; Forest Service Road 615a, 30 Apr and 4 May 1996, D.J. Wright.

3550 *Acleris youngana* (McDunnough)
Harlan Co., summit of Big Black Mountain, 12 Jul 1980, L.D. Gibson.

CRAMBIDAE

4754 *Synclita tinealis* Munroe
Henderson Co., Frank Sauerheber Unit, Sloughs Wildlife Management Area, 22 Aug 1992, L.D. Gibson.

4769 *Neargyractis slossonalis* (Dyar)
Hopkins Co., 2 miles SE Dawson Springs along Caney Creek, 20 Aug 1999 in light trap, L.D. Gibson. This record is a significant northern range extension. Munroe (1972, p. 116) had records only from Florida but believed it would be recorded in neighboring states.

4981 *Helvibotys pseudohelvialis* (Capps)
Fulton Co., Willingham Bottoms, Rt. 94, 2.5 miles E of Cayce, 8 Sep 1991.

5034 *Pyrausta signatalis* (Wlk.)
Laurel Co., Daniel Boone National Forest, powerline corridor east side of south end of Forest Service Road 775, 30 May, 27 Jun, and 10 Jul 1997 at UV and MV lights near *Monarda* (host plant), L.D. Gibson.

5173 *Diasemiodes nigralis* (Fernald)
Bath Co., Cave Run Recreation Area, Forest Service Road 918, 5 Sep 1987, D.J. Wright.

5248 *Lygropia tripunctata* (Fabricius)
Henderson Co., Frank Sauerheber Unit, Sloughs National Wildlife Area, 10 Sep 1983, C.V. Covell Jr.

5450 *Parapediasia decorella* (Zinck.)
Powell Co., Natural Bridge State Park, 7 Jul 1981, in light trap, C.C. Cornett.

5653 *Acrobasis vaccinii* Riley
Laurel Co., junction of Forest Service roads 121 and 4158; 18 May 1996; Forest Service Road 131, 30 May 1992; D.J. Wright.

5766 *Immyria nigrovittella* Dyar
Laurel Co., Daniel Boone National Forest, Forest Service Road 615a, 22 Apr 1995 and 4 May 1996, D.J. Wright; powerline corridor E side of S end of State Road 775, 30 May 1997, L.D. Gibson.

5775 *Salebriaria tenebrosella* (Hulst)
Rowan Co., County Road 1274, 2 miles W of Rt. 519, 16 Jul 1994, L.D. Gibson; Rowan Co., 3.3 miles S of Rt. 519, and also east end of Clack Mtn. Rd. West, 26 Aug 1994, D.J. Wright.

5777.1 *Salebriaria atratella* Blanchard & Knudson
Laurel Co., Bolton Branch, 18 May 1996, D.J. Wright.

5794 *Nephoterix vetustella* (Dyar)
Boone Co., Big Bone Lick State Park, 5 May 1980; Powell Co., Tunnel Ridge, Red River Gorge

- Geological Area, 19 Jun 1993; both collected by L.D. Gibson.
- 5806 *Nephoterix crassifasciella* Ragonot
Laurel Co., Daniel Boone National Forest, powerline corridor, E side of S end of State Road 775, 27 Jun and 10 Jul 1997, L.D. Gibson.
- 5944 *Homoeosoma deceptorium* Heinrich
Boone Co., Camp Ernst, 17 Aug 1979, L.D. Gibson.
- 5953 *Laetilia fiskeella* Dyar
Laurel Co., Daniel Boone National Forest, powerline corridor, E side of S end of State Road 775, 17 May and 27 Jun 1997 at lights, L.D. Gibson.
- 6122 *Stenoptilodes brevipennis* (Zeller)
Bullitt Co., Bernheim Arboretum and Research Forest, 20–24 Apr 1976 in Malaise trap, A.J. Brownell.

PTEROPHORIDAE

- 6107 *Gillmeria pallidactyla* (Haworth)
Harlan Co., summit of Big Black Mountain, 28 Jun 1999, male and female, Reed A. Watkins.
- 6168 *Oidaematophorus eupatorii* (Fernald)
Harlan Co., Big Black Mountain, 14 Jul 1979; Pine Mountain Settlement School, 4 Jul 1977; both collected at black light by C.V. Covell Jr.

GEOMETRIDAE

- 6851 *Philtraea monillata* Buckett
Carlisle Co., Sandy Branch, Burkley, 4 Sep 1999, numerous specimens in light traps, C.V. Covell Jr., L.D. Gibson, and others.

NOCTUIDAE

- 8658 *Selenisa sueroides* (Guenée)
Carlisle Co., Sandy Branch, 10 Oct 1999, in light trap, William R. Black Jr.
- 9386 *Luperina trigona* Smith
Larvae were collected from *Arundinaria* (cane) stalks by W.R. Black Jr. and Eric Quinter 10–18 May 1999 in the following localities: Ballard Co., Stovall Creek; Carlisle Co., Sandy Branch near Burkley; Fulton Co., Willingham Bottoms, Rt. 94, 2.5 miles E of Cayce and Reelfoot Lake National Wildlife Area; Graves Co., Boaz; Livingston Co., Burna; McCracken Co., Massac Creek Bottoms, Paducah. An adult was collected in a light trap in

- McCracken Co., 1 mile W of Clinton Rd., 4 Sep 1999 by W.R. Black Jr.
- 9329.1 *Apamea* undescribed species K. Mikkola
Bullitt Co., Bernheim Arboretum and Research Forest, 12–18 Jul 1976 in Malaise trap, A.J. Brownell; Jefferson Co., Valley Station, “Aug.”, S. Scholz; Russell Co., Lake Cumberland State Park, 11 Jun 1980, C.C. Cornett.
- 9419 *Oligia mactata* (Guenée)
Jefferson Co., Valley Station, 26, 29 Sep, 2–5 and 20–29 Oct 1997, S. Scholz.
- 9425 *Meropleon cosmion* Dyar
McCracken Co., Paducah, Massac Creek Bottoms in cane, 13 Nov 1999 in light trap, W.R. Black Jr.
- 9681.1 *Elaphria cornutinis* Saluke & Pogue.
Two male paratypes were listed from Kentucky by Saluke and Pogue (2000): Metcalfe Co., Highway 218 north of Center, 25 Apr and 6 May 1994, C. Cook. This species is very similar to *E. festivoides* (Gn.). Some records listed under *E. festivoides* in Covell (1999, p. 160) will turn out to be this newly described species.
- 10460 *Leucania calidior* (Forbes)
Carlisle Co., Sandy Branch near Burkley, 2 Oct 1999, in light trap, W.R. Black Jr.; Livingston Co., Burna, 10–18 May 1999, larva in cane reared to adult, E.L. Quinter and W.R. Black Jr. The species name is misspelled as “callidior” in Hodges et al. (1983) (fide E.L. Quinter).

LITERATURE CITED

- Covell, C. V., Jr. 1984. A field guide to the moths of eastern North America. Houghton Mifflin, Boston, MA.
- Covell, C. V., Jr. 1999. The butterflies and moths (Lepidoptera) of Kentucky: an annotated checklist. Kentucky State Nat. Preserves Comm. Tech. Ser. 6.
- Hodges, R. W. 1999. The moths of America north of Mexico. Fasc. 7.6 Gelechioidea: Gelechiidae (part). Wedge Entomol. Res. Found., Washington, DC.
- Hodges, R. W. et al. 1983. Check list of the Lepidoptera of America North of Mexico. Wedge Entomol. Res. Found., Washington, DC.
- Munroe, E. G. 1972. The moths of America north of Mexico. Fasc 13.1A. Pyraloidea: Pyralidae (Part). Wedge Entomol. Res. Found., Washington, DC.
- Saluke, S. V., and M. G. Pogue. 2000. Resolution of the *Elaphria festivoides* (Guenée) species complex (Lepidoptera: Noctuidae). Proc. Entomol. Soc. Washington 102:233–270.

Comparative Effects of Zinc, Lead, and Cadmium on Body and Tissue Weights of Weanling, Adult, and Aged Rats

F.N. Bebe and Myna Panemangalore¹

Nutrition and Health Program, Kentucky State University, Frankfort, Kentucky 40601

ABSTRACT

The effects of feeding various levels of Zn in the diet and Pb and Cd in drinking water were determined in weanling, adult, and aged rats for 4 or 8 weeks. Zn levels in the diet were: Zn-deficient diet or Zn0; 60 mg/kg high Zn diet (Zn60) for experiment 1 & 3; 4 mg/kg low Zn diet (Zn4); 24 mg/kg high Zn diet (Zn24) for experiment 2; and 12 mg/kg Zn diet as control. Zn0 diets reduced feed intake (FI) and body weight (BW) in weanling rats by 35% and 80% and BW in aged rats by 60%; BW of Zn4 adult rats decreased by 40% ($P \leq 0.05$). Feed intake of adult and aged rats was comparable among all groups. Feed efficiency (FE) decreased fourfold and twofold in weanling and adult rats, respectively. Liver and kidney weights were significantly lower in weanling group fed Zn0 diet as compared to the control or Zn60 groups ($P \leq 0.05$). Weanling and adult rats given 20 mg/liter Pb and 5 mg/liter Cd in drinking water had lower water intake (WI) than the control ($P \leq 0.05$). These results indicate that Zn0 diet decreased FI, BW, and FE in weanling rats; Zn0 and Zn4 diets reduced BW in adult and aged rats but without a decline in FI. Lead and Cd in drinking water did not affect growth or FI but decreased WI in weanling and adult rats. Although lead and cadmium did not modify growth in either zinc-deficient or low zinc-fed rats, these diets decreased growth and feed efficiency, which may partly be attributed to loss of appetite and altered Zn homeostasis.

INTRODUCTION

Trace elements have an important and critical role in maintaining nutrition and normal health in animals. Zinc is essential for reproduction, growth, and development. As a component of over 200 enzymes, it plays a vital role in cell replication and differentiation and in the structure and function of cellular membranes. The metabolic and physiological status of zinc could be modified or exacerbated by the ingestion of toxic metals such as lead or cadmium, which are widespread contaminants of the food chain and potable water sources (Underwood 1979). The cumulative retention of lead and cadmium after chronic low-level exposure can result in manifestations of toxicity such as disturbance of heme synthesis, dysfunction of the nervous system, and renal damage (Friberg et al. 1985). Concurrent exposure to lead and cadmium may increase toxicity of one or both metals and produce significant changes in the metabolism of zinc (Mahaffey et al. 1982).

Coppen-Jaeger and Wilhelm (1989) reported inhibition of zinc absorption in isolated rat intestinal preparations after low-level cadmium exposure. While high levels of zinc may

reduce some of the toxic effects of cadmium (Sato and Nagai 1989), zinc deficiency may enhance toxic effects of even low levels of cadmium (Kunifuji et al. 1987; Tanaka et al. 1995). Zinc is reported to have a protective effect on lead toxicity when both are present in the diet at high levels due to inhibition of lead absorption (Cerklewski and Forbes 1975). Age may affect zinc metabolism because related changes in body composition are often paralleled by a decline in physiological and metabolic functions, which could also modify growth and food conversion efficiency (Sandstead et al. 1982). Age-related differences in response of rats to lead or cadmium exposure were reported by Cory-Slechta et al. (1989) and Song et al. (1986). However, most published research has been done with pharmacological or toxic doses of lead or cadmium using either oral (Sato and Nagai 1989) or parenteral methods (Kunifuji et al. 1987) of exposure, which have little bearing on exposure conditions in the environment and may not be of much practical significance (Sabbioni et al. 1978). Therefore, we hypothesize that low-level exposure to lead and cadmium would not alter zinc absorption and metabolism. To test this hypothesis, we have determined the interaction among lead, cadmium, and zinc in

¹ To whom correspondence should be addressed.

growing, adult, and aged rats fed increasing levels of zinc in the diet.

In this paper, we present data on the interaction of low-level lead and cadmium in drinking water on growth, feed intake, feed efficiency, and tissue weights of weanling, adult, and aged rats fed different levels of zinc in the diet.

MATERIALS AND METHODS

Male Sprague Dawley rats (Harlan Sprague Dawley, Inc.) 27 days, 8 weeks, or 18 months old were housed singly in suspended polypropylene cages on stainless steel racks in temperature controlled rooms with 12 h light/dark cycles. During a 1-week period of acclimation, rats were fed the control diet and given distilled drinking water. The experimental diets (Harlan Teklad, Madison, WI) containing different levels of zinc were based on modifications of the zinc-deficient diet (AIN93 formula), using egg white as the protein source and a mixture of starch and dextrin as the carbohydrate source (Reeves et al. 1993). Zinc sulfate was added to experimental diets as follows: Control group = 12 mg/kg (Zn12); zinc-deficient group = 0 mg/kg (Zn0); low-zinc group = 4 mg/kg (Zn4); and high-zinc group = 24 mg/kg (Zn24) or 60 mg/kg (Zn60). All diets were analyzed for their zinc content by atomic absorption spectrophotometry, which was within a 5% range of the expected level. Zinc content of the diets as mg Zn/kg diet were Zn0 = 0.8, Zn4 = 4.9, Zn12 = 12.7, Zn24 = 25.0, and Zn60 = 60.5.

After the acclimation period, the rats were randomized and grouped such that the average initial body weights were similar in all groups (six rats/group; the deficient and pair-fed groups had weight-matched pairs) and were assigned to the experimental diets described above. The rats were given distilled water containing 10 mg/liter sodium as NaCl, or 20 mg/liter Pb as lead acetate, or 5 mg/liter Cd as cadmium chloride (0 = control). Rats had free access to feed and water as follows: experiment 1 and 3, 30 days and experiment 2, 60 days. In experiment 1 (72 weanling) and experiment 3 (36 aged) rats were fed diets Zn0, Zn12, Zn60, or Zn12PF (pair-fed control diet daily to the intake of the Zn0 group); in experiment 2 (72 adult) rats were fed diets Zn4, Zn12, Zn24, or Zn12PF (pair-fed control

diet daily to the intake of the Zn4 group). Feed intake (FI) was determined daily, whereas body weight (BW) and water intake (WI) were recorded weekly. Feed efficiency (FE) was calculated as the ratio of FI to weight gain (WG). The three experiments were conducted separately under uniform conditions. For ease of handling 72 rats, experiments 1 and 2 were split in half such that each group had 3 rats, and each batch was run under identical conditions, 1 week apart. The procedures for these animal experiments were approved by the Kentucky State University Animal and Human Welfare Committee.

At the end of the experimental period, rats were terminated under diethyl ether anesthesia for removal of liver and kidneys. The organs were cleansed of extraneous tissue, weighed, and stored at -70°C . All data were statistically analyzed by ANOVA using the SAS program and a two-way, 4 (Zn levels) \times 3 (Na/Pb/Cd levels) factorial design. Significant differences between means were obtained by the Duncan's multiple range test.

RESULTS

The general appearance of the animals was influenced by dietary zinc. Weanling rats were especially affected by zinc deficiency as they lost abdominal hair and had hairless patches elsewhere on the body; those fed Zn12 and Zn60 had smooth hair. Adult rats fed low zinc diet (Zn4) did not lose hair but the texture of the hair was rough. No physical difference in appearance was noted in aged rats. Growth, FI, FE, and tissue weight data are presented here.

Since exposure to oral lead and cadmium did not influence BW or FI, all data presented in Figure 1 were merged ignoring lead and cadmium exposure ($n = 18$). Weanling rats fed Zn0 and Zn12PF diets had significantly lower FI (Figure 1A), weight gain (WG) (Figure 1B), and FE as compared to control (Figure 1C; $P \leq 0.05$): Feed intake was about 35% lower, while WG was reduced by 80% and 54%, respectively. Weanling rats fed diet Zn60 grew throughout the 4-week experiment and their FI, WG, and FE were comparable to those of the control group. Feed efficiency was lower among rats fed Zn0 and Zn12PF, and both were two-fold and four-fold lower than the control. Growth and FI data of adult

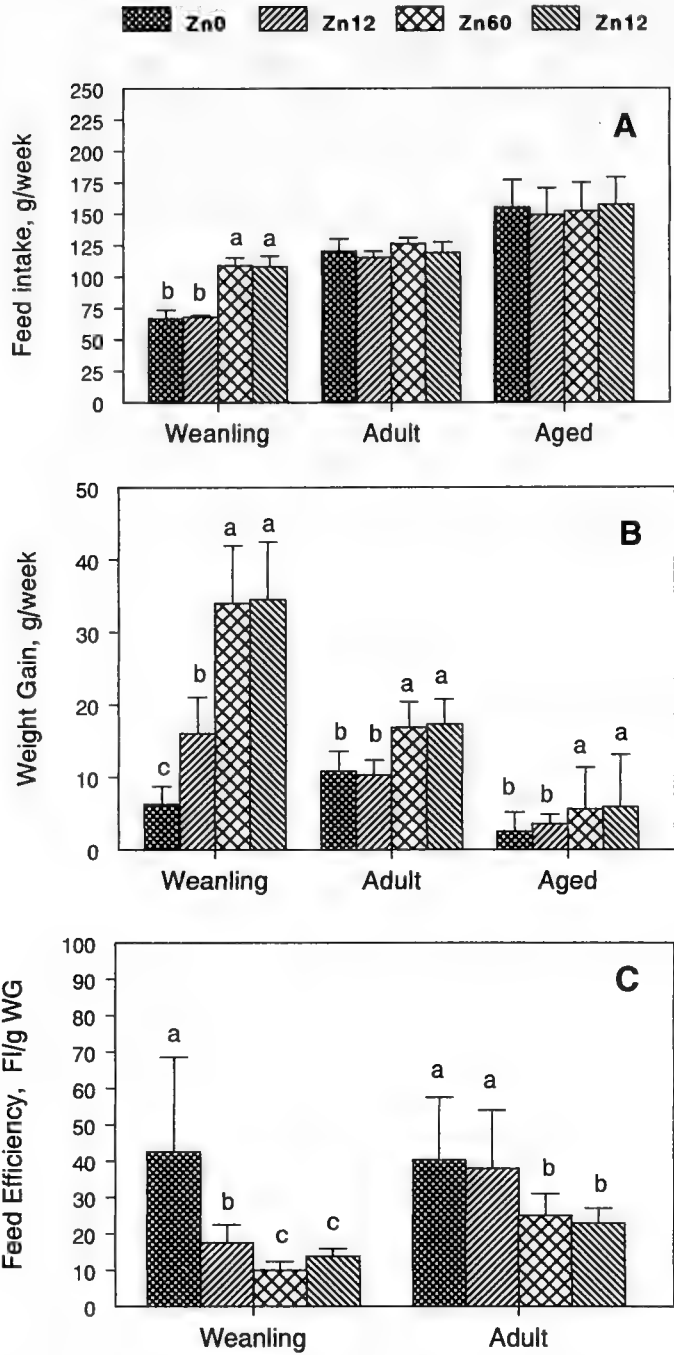


Figure 1. Feed intake (A), weight gain (B), and feed efficiency (C) of weanling, adult, and aged rats fed Zn0 (Zn4 for adult rats), Zn12PF, Zn60, and Zn12 (control) for 4 weeks (8 weeks for adult rats). Means \pm SD; $n = 18$ (weanling and adult groups). Means \pm SD; $n = 9$ (aged groups). Means in different diet groups not sharing the same superscript are significantly different at ($P \leq 0.05$).

rats fed low and high zinc diets for 8 weeks are presented in Figure 1A, 1B, 1C. Weight gain and FE of rats fed Zn4 and Zn12PF diets were 40% and two-fold lower than those fed the control diet ($P \leq 0.05$), respectively. There was no significant difference in FI among all the groups ($P \leq 0.05$). Aged rats showed a similar BW and FI pattern as adult rats (Figure 1A, 1B, 1C). Feed intake was comparable

among aged rats in the different diet groups irrespective of zinc level, but WG was 60% and 43% lower in Zn0 and Zn12PF groups than the control or Zn60 group ($P \leq 0.05$). Rats (especially the Zn0 fed groups) experienced a day-to-day increase or decrease in feed consumption, which indicated cycling of feed intake. Due to wide variation in FI, FE values of aged rats could not be calculated accurately and therefore are not reported here. Low-level oral exposure to lead or cadmium had no effect on growth and FI of weanling, adult, or aged rats.

Tissue weights of rats are presented in Table 1. In weanling rats (experiment 1), average liver weights were about 54% lower in the Zn0 and Zn12PF groups than in the control or Zn60 groups ($P \leq 0.05$). Similarly, kidney weights were a third lower in the Zn0 and Zn12PF groups compared to the control ($P \leq 0.05$). Also, mean liver and kidney wet weight/body weight ratios were comparable among all dietary groups. Such differences in tissue weights were not observed in adult or aged rats. Lead and cadmium exposure had no effect on tissue weights.

Water intake data of rats for experiments 1 and 2 are presented in Table 2. There was significant interaction between dietary zinc levels and the concentration of lead and cadmium in drinking water, especially in the zinc-deficient group. Water intake was significantly lower in weanling and adult rats given lead and cadmium in drinking water than those given sodium in water (control), regardless of the level of zinc in the diet ($P \leq 0.05$). The data also indicated that, in weanling rats, WI was significantly influenced not only by the presence of lead and cadmium in drinking water but also by the amount of zinc in the diet. Water intake for experiment 3 could not be accurately measured because of excessive spillage, so these data are not presented.

DISCUSSION

Cellular homeostasis, a major regulator of zinc metabolism, ensures that over a wide range of zinc intake, tissue or cellular zinc levels are maintained at physiological concentrations by either enhancing absorption or decreasing loss through the gastrointestinal tract (Golden 1989). In our study, we found that Zn0 diet significantly decreased feed intake,

Table 1. Tissue weights of weanling (n = 18), and aged (n = 9) rats fed zinc deficient (Zn0), high zinc (Zn60), or control (Zn12) zinc diets for 4 weeks, and adult rats (n = 18) fed low zinc (Zn4), high zinc (Zn24), and control zinc (Zn12) for 8 weeks. Data are presented as Mean ± SD.

Parameters	Experiment	Zinc levels in the diet			
		Zn 0	Zn12PF ¹	Zn 60	Zn12 ¹
Liver, g	1 (W) ²	5.4 ± 0.8 ^b	5.1 ± 1.1 ^b	9.5 ± 1.5 ^a	9.7 ± 1.3 ^a
	2 (A)	11.2 ± 1.5	10.4 ± 1.1	11.6 ± 1.5	11.8 ± 1.5
	3 (AG)	17.8 ± 2.3	16.8 ± 2.4	17.9 ± 0.8	17.2 ± 2.5
Kidney, g	1 (W) ²	1.2 ± 0.1 ^b	1.3 ± 0.2 ^b	1.9 ± 0.2 ^a	1.9 ± 0.2 ^a
	2 (A)	2.2 ± 0.1	2.1 ± 0.2	2.2 ± 0.02	2.2 ± 0.2
	3 (AG)	3.9 ± 0.9	3.1 ± 0.5	3.2 ± 0.01	3.2 ± 0.4
Liver wt/body wt ratio	1 (W) ²	0.04 ± 0.007	0.03 ± 0.005	0.04 ± 0.002	0.04 ± 0.003
	2 (A)	0.03 ± 0.004	0.03 ± 0.002	0.03 ± 0.002	0.03 ± 0.003
	3 (AG)	0.04 ± 0.005	0.03 ± 0.005	0.04 ± 0.003	0.03 ± 0.004
Kidney wt/body wt ratio	1 (W) ²	0.01 ± 0.001	0.01 ± 0.001	0.01 ± 0.001	0.01 ± 0.001
	2 (A)	0.01 ± 0.001	0.01 ± 0.005	0.01 ± 0.004	0.01 ± 0.003
	3 (AG)	0.01 ± 0.003	0.01 ± 0.001	0.01 ± 0.001	0.01 ± 0.001

Data were grouped ignoring Pb, Cd, exposure as no significant differences were observed between these groups. Means in rows with letter superscripts indicate significant differences; absence of letters indicates means are not significantly different at P ≤ 0.05.
¹ Rats in pair-fed (Zn12PF) control group were fed control diet (Zn12) in the amount equal to that eaten by either Zn0 or Zn4 groups.
² W = weanling; A = Adult; AG = Aged rats (see materials and methods for details).

body weights, and feed conversion efficiency of weanling rats and that long-term (8 week) Zn4 feeding also reduced body weights without a concomitant decline in FI, which suggests alteration of zinc homeostasis or a decrease in the availability of zinc for growth and maintenance. While exposure to low levels of lead and cadmium did not affect FI or BW in any age group, it decreased WI significantly in weanling and adult rats.

Guigliano and Millward (1984) reported that in zinc deficient animals, zinc may be redistributed from bone and conserved in muscles, being released only for the growth and vital functioning of essential tissues in a catabolic state. It is not clear if the failure of zinc-

deprived rats to grow is a result of zinc on appetite or an effect of impaired cell division on growth (O'Dell and Reeves 1989). Rains and Shay (1994) initially suggested that reduced FI in Zn deficient rats could be because of impaired function of neuropeptide Y, a known stimulator of appetite and carbohydrate consumption. More recently, neuropeptide Y has been shown to increase in zinc deficiency, probably to restore normal FI, but this is prevented by receptor binding of this peptide (Lee et al. 1998). Our data suggest that reduction in growth could be due to reduced FI as well as impaired cell division chiefly in weanling rat, since pair-fed groups lost less weight than did the Zn0 (weanling) and

Table 2. Water intake of weaning rats fed zinc-deficient (Zn0) or high zinc (Zn60) diets for 4 weeks, or adult rats fed low zinc (Zn4) or high zinc (Zn24) diets for 8 weeks, and exposed to lead or cadmium in drinking water. Means ± SD; n = 18/group.

Experiment	Metals in water	Zinc levels in the diet (ml water intake/wk)			
		Zn 0	Zn 12 PF ¹	Zn 60	Zn12 ¹
1 (W) ²	Na	130 ± 7 ^{a,f}	140 ± 28 ^{a,ef}	161 ± 13 ^{a,e}	178 ± 16 ^{a,d}
	Pb	100 ± 22 ^{b,f}	131 ± 10 ^{b,e}	153 ± 9 ^{b,d}	162 ± 15 ^{b,d}
	Cd	89 ± 20 ^{c,f}	118 ± 16 ^{c,e}	150 ± 13 ^{b,d}	152 ± 14 ^{b,d}
2 (A) ²	Na	180 ± 27 ^a	167 ± 35 ^a	160 ± 15 ^a	166 ± 20 ^a
	Pb	154 ± 20 ^b	149 ± 20 ^b	160 ± 11 ^a	148 ± 11 ^a
	Cd	150 ± 25 ^b	144 ± 20 ^b	130 ± 15 ^b	144 ± 13 ^b

ANOVA indicates interaction between zinc levels, and lead and cadmium in water. a, b, c, denote significant differences in each row; d, e, f, denote significant differences in each column (P ≤ 0.05), within each experiment.
¹ Rats in pair-fed (Zn12PF) control group were fed control diet (Zn12) in the amount equal to the intake by either Zn0 or Zn4 groups. (See materials and methods for details).
² W = weanling; A = Adult. Data for Experiment 3 are not available.

Zn4 (adult) rats, respectively. Thus, the higher weight loss in the Zn0 group may be attributed partly to zinc deprivation and lowered FI; that in the pair-fed group, to lowered FI.

Numerous reports (Abdel-Mageed and Oehme 1991; Faraji and Swendseid 1983; Guigliano and Millward 1984) attest to the significant decline of FI and growth in Zn0 weanling rats and support our observations. In our study, aged rats on Zn0 diet for 4 weeks also lost weight but, unlike weanling and adult rats, without a significant decrease in FI. One reason for this may be the variation and cycling of FI we found in all groups of aged rats irrespective of zinc level in the diet; whereas, cycling of FI was found only in zinc-deficient weanling and adult rats. However, recent studies analyzing food-intake patterns of zinc deficient rats have shown that the characteristic cyclical variation in FI and body weight changes were found not only by group but also in each individual rat fed zinc-deficient diets (Tamaki et al. 1995) and that body weight change is generally well synchronized with that of FI (Aiba et al. 1997). The reduction in feed conversion efficiency we found in weanling rats was reported previously by William and Mills (1970). Conflicting reports in the literature suggest that zinc levels as high as 1000 $\mu\text{g/g}$ diet fed for 8 weeks do not alter FI or growth, while zinc levels up to 2500 $\mu\text{g/g}$ diet for 3 weeks decrease growth (Abdel-Mageed and Oehme 1991; Song et al. 1986; Story and Greger 1987). Toxicity of such high zinc levels may have influenced their results. Our results support those of Abdel-Mageed and Oehme (1991), since moderately high zinc (60 $\mu\text{g/g}$) diets did not produce any change in growth or FI.

In our study, low levels of lead and cadmium in drinking water did not alter feed intake or body weight when dietary zinc levels were deficient or low or when the diets contained high zinc levels, indicating a lack of interaction between zinc and toxic metals. Previous reports have shown that high levels of lead and cadmium administered through drinking water or parenterally reduce zinc absorption and thereby body weight (Kunifuji et al. 1987; Tanaka et al. 1995) or increase the toxic effect of cadmium (Sato and Nagai 1989). High zinc intake is known to protect against toxic effects of lead (Cerklewski and Forbes 1975). In rats

fed normal zinc diets, Kirchgessner et al. (1987) found that age did not modify the effects of increasing levels of lead or cadmium in the diet. This report supports our observations that at low levels of lead or cadmium exposure there is no interaction between dietary zinc levels or age of the animal on body weight and feed intake.

Although organ weight is known to decline with BW, higher organ to BW ratios in weanling rats fed zinc-deficient diets have been reported (Abdel-Mageed and Oehme 1991; Meydani et al. 1983), but our data do not completely agree with these observations since we found that, in weanling rats, liver and kidney weights were significantly lower in Zn0 or pair-fed groups as compared to the controls. This parallel and proportional decrease or increase in organ and body weights is indicated by the similar kidney and body weight ratios of all groups in the three experiments irrespective of age; it refutes the effect of zinc deficiency on decline of only skeletal mass suggested by Abdel-Mageed and Oehme (1991) who did not include a pair-fed group for comparison. Our observation that lead and cadmium had no significant effect on organ weights in all age groups is similar to reports by Rader et al. (1981), Vyskocil et al. (1991), and Cory-Slechta et al. (1989) in weanling, adult, and aged rats, respectively. Age-related changes begin to accelerate in rodents after 16 months of age (Cory-Slechta et al. 1989). The aged rats in our study were 18 months old and yet showed no adverse effects of short-term exposure to low lead and cadmium levels.

Addition of low levels of toxic metals in drinking water is assumed not to alter WI, but our data demonstrate that WI decreased significantly in both weanling and adult rats when low levels of lead and cadmium salts were added to drinking water. These data are largely supported by the reports of Vyskocil et al. (1990) in weanling rats, Kotsonis and Klaassen (1978) in adult rats, and Cory-Slechta (1990) in aged rats using much higher (10–100 times) concentrations of lead or cadmium than we used. We were unable to measure WI accurately in aged rats; thus the decrease in WI in this group cannot be confirmed. It is apparent that reduced FI produced by Zn0 diet also significantly decreased WI in weanling rats, while increasing Zn in the diet did not alter WI. We

found no such changes in adult or aged rats, indicating a direct relationship between FI and WI in weanling rats.

From our data, it is apparent that zinc-deficient or low-zinc diets fed for a prolonged period can significantly decrease weight gain and feed conversion efficiency in weanling and adult rats, respectively. These effects may be attributed to impaired cell division and metabolism caused by altered Zn homeostasis due to Zn deficiency, as well as reduced FI resulting from loss of appetite. Low-level oral lead and cadmium exposure was not toxic to all age groups of rats. Significant reduction in water intake after addition of lead or cadmium suggests that this may not be an appropriate method of oral exposure when quantitative toxic metal exposure is critical, as actual intake may vary significantly within and between groups. Addition of toxic metals to the diet may perhaps be a preferable method.

ACKNOWLEDGMENTS

We thank Joyce Owens (Animal Facilities manager), and students K. Mahal, A. Mutiso, and C. Moore for assisting with animal experiments. This research was funded by USDA Grant 90-38814-5540.

LITERATURE CITED

- Abdel-Mageed, A.B., and F.W. Oehme. 1991. The effect of various dietary zinc concentrations on the biological interactions of zinc, copper and iron in rats. *Biol. Trace Elem. Res.* 29:239–256.
- Aiba, K., M. Kimura, S. Sakata, K. Matsuda, M. Kaneko, S. Onosaka, Y. Yamaoka, and N. Tamaki. 1997. Cosinor analysis of feed intake cycle of rats fed a zinc deficient diet and the effect of zinc supplementation. *J. Nutr. Sci. Vitaminol.* 43:327–343.
- Bebe, F.N., and M. Panemangalore. 1996. Modulation of tissue trace metal concentrations in weanling rats fed different levels of zinc and exposed to oral lead and cadmium. *Nutr. Res.* 16:1369–1380.
- Cerklewski, F.L., and R.M. Forbes. 1975. Influence of dietary zinc on lead absorption in the rat. *J. Nutr.* 106: 689–696.
- Coppen-Jaeger, D.E., and M. Wilhelm. 1989. The effects of cadmium on zinc absorption in isolated rat intestinal preparations. *Biol. Trace Elem. Res.* 21:207–212.
- Cory-Slechta, D.A. 1990. Alterations in tissue lead distribution and hematopoietic indices during advanced age. *Arch. Toxicol.* 64:31–37.
- Cory-Slechta, D.A., B. Weiss, and C. Cox. 1989. Tissue distribution of lead in adult vs old rats: a pilot study. *Toxicology* 59:139–150.
- Faraji, B., and M.E. Swendseid. 1983. Growth rate, tissue zinc levels and activities of selected enzymes in rats fed a zinc deficient diet by gastric tube. *J. Nutr.* 113:447–455.
- Friberg, L., C.G. Elinder, T. Kjellstrom, and G. Nordberg. 1985. Cadmium and health. CRC Press, Boca Raton, FL.
- Golden, M.H.N. 1989. Diagnosis of zinc deficiency. Pages 323–332 in C.F. Mills (ed). *Zinc in human biology*. Springer-Verlag, London, U.K.
- Guigliano, R., and D.J. Millward. 1984. Growth and zinc homeostasis in the severely zinc-deficient rat. *British J. Nutr.* 52:545–560.
- Kirchgessner, M., A.M. Reichlmayr-Lais, and K. Stockl. 1987. Retention of lead in growing and adult rats on varying lead supply. Pages 689–690 in L.S. Hurley, C.L. Keen, B. Lonnerdal, and R.B. Rucker (eds). *Trace elements in animals and man*. Plenum, New York, NY.
- Kotsonis, F.N., and C.D. Klaassen. 1978. The relationship of metallothionein to the toxicity of cadmium after prolonged oral administration to rats. *Toxicol. Appl. Pharmacol.* 46:39–54.
- Kunifujii, Y., T. Nakamura, and M. Takasugi. 1987. Influence of cadmium in the distribution of essential trace elements in the liver and kidneys of rats. *Biol. Trace Elem. Res.* 14:237–248.
- Lee, R.G., T.M. Rains, C. Towar-Palacio, J.L. Beverly, and N.F. Shay. 1998. Zinc deficiency increases hypothalamic neuropeptide Y and neuropeptide Y mRNA levels and does not block neuropeptide Y-induced feeding in rats. *J. Nutr.* 128:1218–1223.
- Mahaffey, K.R., S.G. Caper, B.C. Gladen, and B.A. Fowler. 1982. Concurrent exposure to lead, cadmium and arsenic. Effect on toxicity and tissue metal concentrations in the rat. *J. Lab. Clin. Med.* 66:463–481.
- Meydani, S.N., M. Meydani, and J. Dupont. 1983. Effect of prostaglandin modifiers and zinc deficiency on possibly related functions in rats. *J. Nutr.* 113:494–500.
- O'Dell, B.L., and P.G. Reeves. 1989. Zinc status and food intake. Pages 173–179 in C.F. Mills (ed). *Zinc in human biology*. Springer-Verlag, London, U.K.
- Panemangalore, M. 1993. Interaction among zinc, copper and cadmium in rats: effect of low zinc and copper diets and oral cadmium exposure. *J. Trace Elem. Exp. Med.* 6:125–139.
- Rader, J.I., J.T. Peeler, and K.R. Mahaffey. 1981. Comparative toxicity and tissue distribution of lead acetate in weanling and adult rats. *Environ. Health Perspect.* 42:187–195.
- Rains, T.M., and N.F. Shay. 1994. Macronutrient preference in the rat during zinc deficiency-induced anorexia. *FASEB J.* 8:A730.
- Reeves, P.G., F.H. Nielsen, and G.C. Fahey, Jr. 1993. AIN 93 purified diets for laboratory rodents: final report of the American Institute of Nutrition ad hoc writing committee on reformulation of AIN 76 rodent diet. *J. Nutr.* 123:1939–1951.
- Sabbioni, E., E. Marafante, L. Amantini, L. Ubertalli, and

- R. Pietra. 1978. Cadmium toxicity under long term low level exposure (LLE) conditions. Metabolic patterns in rats exposed to present environmental dietary levels of cadmium for two years. *Sci. Total Environ.* 10:135–161.
- Sandstead, H.H., L.K. Henrickson, J.L. Greger, A.S. Prasad, and R.A. Good. 1982. Zinc nutriture in elderly in relation to taste acuity, immune response and wound healing. *Am. J. Clin. Nutr.* 36:1046–1059.
- Sato, M., and Y. Nagai. 1989. Effect of zinc deficiency on the accumulation of metallothionein and cadmium in the rat liver and kidney. *Arch. Environ. Contam. Toxicol.* 18:587–593.
- Song, M.K., N.F. Adham, and M.E. Ament. 1986. Levels and distribution of zinc, copper, magnesium and cadmium in rats fed different levels of dietary zinc. *Biol. Trace Elem. Res.* 11:75–88.
- Story, M.L., and J.L. Greger. 1987. Iron, zinc and copper interactions: chronic versus acute responses of rats. *J. Nutr.* 117:1434–1442.
- Tamaki, N., S. Fujimoto-Sakata, M. Kikugawa, M. Kaneko, S. Onosaka, and T. Takagi. 1995. Analysis of cyclic feed intake in rats fed on a zinc deficient diet and the level of dihydropyrimidinase. *Brit. J. Nutr.* 73:711–722.
- Tanaka, M., M. Yanagi, K. Shiota, Y. Une, Y. Nomura, T. Masaoka, and F. Akahori. 1995. Effect of cadmium in the zinc deficient rat. *Vet. Human Toxicol.* 37:203–208.
- Underwood, E.J. 1979. Environmental sources of heavy metals and toxicity to man. *Prog. Water Technol.* 11:33–45.
- Vyskocil, A., Z. Fiala, V. Lacinova, and E. Ettlerova. 1991. A chronic study of lead acetate in female rats. *J. Appl. Toxicol.* 11:385–386.
- Vyskocil, A., Z. Fiala, E. Ettlerova, and I. TeRejnorova. 1990. Influence of chronic lead exposure on hormone levels in developing rats. *J. Appl. Toxicol.* 10:301–302.
- Williams, R.B., and C.F. Mills. 1970. The experimental production of zinc deficiency in the rat. *Brit. J. Nutr.* 24:989–1003.

Rare and Extirpated Biota of Kentucky

Kentucky State Nature Preserves Commission

801 Schenkel Lane, Frankfort, Kentucky 40601

ABSTRACT

The Kentucky State Nature Preserves Commission has updated and revised the lists of rare and extinct or extirpated biota last published in 1996 and updated in 1997 and 1999. The newly revised lists include a lichen, and 389 plant and 282 animal taxa considered rare in Kentucky, and 4 plant and 42 animal taxa considered extirpated from Kentucky or extinct.

INTRODUCTION

The Kentucky State Nature Preserves Commission (KSNPC) is mandated to identify and protect natural areas to conserve Kentucky's natural heritage. To accomplish this mandate KSNPC works in cooperation with many scientific authorities in the public, private, and academic sectors. To help focus its conservation activities, KSNPC has developed a list of taxa native to the state that are considered rare. A list of species presumed extinct or extirpated from Kentucky is also maintained to document the loss of biodiversity, much of which is attributable to human activities. The overall goal of publishing these lists is to assist in the recovery and preservation of Kentucky's rich natural diversity.

KSNPC uses The Nature Conservancy's standardized Natural Heritage Program (NHP) methodology (TNC 1988) to manage distributional and ecological information on rare taxa, high quality natural communities, and other unique natural features in map, manual, and computer files. This information is used to locate aggregations of these entities for monitoring and protection. The NHP methodology is well suited for the revision process outlined below.

METHODS

Each taxon listed by KSNPC (1996, 1997, 1999), as well as other unlisted organisms, were evaluated to assign a conservation status. The evaluation criteria used included the number, age, and accuracy of occurrences; historical and present geographic distribution; habitat requirements; threats to the taxon including habitat loss; and ecological fragility. The information used to make the evaluation was that available as of 1 Jan 2000. The re-

sultant list and proposed status designations were submitted to knowledgeable individuals for peer review and suggestions for taxa to add and delete. All comments received were considered and in many cases discussed with the reviewer before the list was finalized.

Sources consulted for the plant and lichen names are: Anderson (1990); Crum et al. (1990); Egan (1987); and Kartesz (1994). The sources consulted for the common and scientific names of animals are as follows: gastropods—Hubricht (1985) and Turgeon et al. (1998); freshwater mussels—Gordon (1995) and Turgeon et al. (1998); crustaceans—Barr (1968), Holsinger (1972), Taylor and Sabaj (1998), USFWS (1994), and Williams et al. (1989); insects—Arnett (1983); Barr (1996), Cassie et al. (1995), Krekeler (1973), McCafferty (1996), Miller (1992), Morse (1993), Paulson and Dunkle (1999), Schuster (1997), and Schweitzer (1989); fishes—Ceas and Page (1997), Page and Burr (1991), Robins et al. (1991), USFWS (2000), and Warren (1992); amphibians and reptiles—Collins (1990), Frost (1985), and King and Burke (1989); breeding birds—AOU (1998); mammals—Hall (1981), Jones et al. (1992), and Wilson and Reeder (1993).

Status Designations

The intent of assigning status designations is to (1) indicate the degree of rarity of the taxon, (2) indicate the degree of threat to the continued survival of the taxon, and (3) aid in establishing conservation priorities. The five KSNPC status designations defined below have no legal or statutory implication.

Endangered (E). A taxon in danger of extirpation and/or extinction throughout all or a significant part of its range in Kentucky.

Threatened (T). A taxon likely to become endangered within the foreseeable future throughout all or a significant part of its range in Kentucky.

Special Concern (S). A taxon that should be monitored because (1) it exists in a limited geographic area in Kentucky, (2) it may become threatened or endangered due to modification or destruction of habitat, (3) certain characteristics or requirements make it especially vulnerable to specific pressures, (4) experienced researchers have identified other factors that may jeopardize it, or (5) it is thought to be rare or declining in Kentucky but insufficient information exists for assignment to the threatened or endangered status categories.

Historical (H). A taxon that has not been reliably reported in Kentucky since 1980 but is not considered extinct or extirpated—see next designation.

Extinct/Extirpated. A taxon for which habitat loss has been pervasive and/or concerted efforts by knowledgeable biologists to collect or observe specimens within appropriate habitat have failed.

Federal statuses (NMFS 1999; USFWS 1999, 2000) are defined below. Non-breeding birds with a federal status occurring as migrants or visitors in Kentucky (e.g., *Charadrius melodus*, *Mycteria americana*) are not included on the list.

Endangered (E). "... any species ... in danger of extinction throughout all or a significant portion of its range ..." (USFWS 1992).

Threatened (T). "... any species ... likely to become an endangered species within the foreseeable future throughout all or a significant portion of its range" (USFWS 1992).

Proposed Endangered (PE). A taxon proposed for listing as endangered.

Candidate (C). Taxa for which the USFWS has "... sufficient information on biological vulnerability and threats to support proposals to list them as endangered or threatened" (USFWS 1999).

DISCUSSION

The list of rare biota includes a lichen and 389 plant and 282 animal taxa considered rare

in Kentucky (Tables 1, 2). Based on generally accepted estimates of the number of native taxa in Kentucky and excluding extinct/extirpated members of each group, the following approximate percent of the groups indicated can be considered Endangered, Threatened, of Special Concern, or Historical: vascular plants—16.5%, gastropods—9.7%, freshwater mussels—42.9%, fishes—26.6%, amphibians and reptiles—28.4%, breeding birds—30.6%, and mammals—21.5%. Although KSNPC continues to refine and expand this list to include new groups, the list does not adequately treat or include several groups of organisms found in Kentucky. The fungi, liverworts, insects, amphipods, isopods, and other groups are important elements of our natural heritage but are poorly known in Kentucky. Researchers are encouraged to continue to gather and publish information about these groups to assist in the evaluation and inclusion of rare taxa on future lists.

Four plants and 42 animals are presumed extinct or extirpated from Kentucky (Tables 2, 3). Most extinct or extirpated animals are freshwater mussels or fishes that have experienced range-wide declines caused by habitat destruction, stream modification, and pollution (Richter et al. 1997). Extirpation and extinction are difficult to prove definitively, so biologists should continue to seek these plants and animals during field activities.

We invite recommendations from knowledgeable individuals regarding native taxa they believe deserve a status change or should be added to or deleted from the list. Each recommendation should include the scientific name of the organism, its habitat requirements, collection information (i.e., localities, number of specimens, dates, disposition of specimens), historical and present distribution, whether the taxon has been specifically sought during field work, threats to its survival, and recommended status. Recommendations should be forwarded to the Director, KSNPC, who will pass the information on to appropriate staff members for timely review and response.

KSNPC intends to publish updated lists in the *Journal* every 4 years. The present lists will be updated annually by submitting a note to the *Journal* listing status and name changes. Interested persons can contact KSNPC for the

Table 1. Endangered, threatened, special concern, and historical biota of Kentucky, 2000.

				Status		
		KSNPC	US	KSNPC	US	
	Lichens					
<i>Phaeophyscia leana</i>		E	—	<i>Ageratina luciae-brauniae</i>	S	—
Bottomland lichen				Lucy Braun's white snakeroot		
	Plants			<i>Agrimonia gryposepala</i>	T	—
				Tall hairy groovebur		
Mosses				<i>Amianthium muscitoxicum</i>	T	—
				Fly-poison		
<i>Abietinella abietina</i>		T	—	<i>Amsonia tabernaemontana</i> var.	T	—
Wire fern moss				<i>gattingeri</i>		
<i>Anomodon rugelii</i>		T	—	Eastern blue-star		
A moss				<i>Anemone canadensis</i>	H	—
<i>Brachythecium populeum</i>		E	—	Canada anemone		
Matted feather moss				<i>Angelica triquinata</i>	E	—
<i>Bryum cyclophyllum</i>		E	—	Filmy angelica		
A moss				<i>Apios priceana</i>	E	T
<i>Bryum miniatum</i>		E	—	Price's potato-bean		
A moss				<i>Arabis hirsuta</i> var. <i>adpressipilis</i>	E	—
<i>Cirriphyllum piliferum</i>		T	—	Hairy rock-cress		
A moss				<i>Arabis missouriensis</i>	E	—
<i>Dicranodontium asperulum</i>		E	—	Missouri rock-cress		
A moss				<i>Arabis perstellata</i>	T	E
<i>Entodon brevisetus</i>		E	—	Braun's rock-cress		
A moss				<i>Aristida ramosissima</i>	H	—
<i>Herzogiella turfacea</i>		E	—	Branched three-awn grass		
A moss				<i>Armoracia lacustris</i>	T	—
<i>Neckera pennata</i>		T	—	Lake cress		
A moss				<i>Aster acuminatus</i>	T	—
<i>Oncophorus raii</i>		E	—	Whorled aster		
A moss				<i>Aster concolor</i>	T	—
<i>Orthotrichum diaphanum</i>		E	—	Eastern silvery aster		
A moss				<i>Aster drummondii</i> var. <i>texanus</i>	T	—
<i>Polytrichum pallidisetum</i>		T	—	Texas aster		
A haircap moss				<i>Aster hemisphericus</i>	E	—
<i>Polytrichum piliferum</i>		E	—	Tennessee aster		
A haircap moss				<i>Aster pilosus</i> var. <i>priceae</i>	T	—
<i>Polytrichum strictum</i>		E	—	White heath aster		
A haircap moss				<i>Aster pratensis</i>	S	—
<i>Sphagnum quinquefarium</i>		E	—	Barrens silky aster		
A peatmoss				<i>Aster radula</i>	E	—
<i>Tortula norvegica</i>		E	—	Low rough aster		
A tortula				<i>Aster saxicastellii</i>	T	—
	Vascular Plants			Rockcastle aster		
<i>Acer spicatum</i>		E	—	<i>Aureolaria patula</i>	S	—
Mountain maple				Spreading false foxglove		
<i>Aconitum uncinatum</i>		T	—	<i>Baptisia australis</i> var. <i>minor</i>	S	—
Blue monkshood				Blue wild indigo		
<i>Adiantum capillus-veneris</i>		T	—	<i>Baptisia bracteata</i> var. <i>leucophaea</i>	S	—
Southern maidenhair fern				Cream wild indigo		
<i>Adlumia fungosa</i>		E	—	<i>Baptisia tinctoria</i>	T	—
Climbing fumitory				Yellow wild indigo		
<i>Aesculus pavia</i>		T	—	<i>Bartonia virginica</i>	T	—
Red buckeye				Yellow screwstem		
<i>Agalinis auriculata</i>		E	—	<i>Berberis canadensis</i>	E	—
Earleaf False Foxglove				American barberry		
<i>Agalinis obtusifolia</i>		E	—	<i>Berchemia scandens</i>	T	—
Ten-lobed false foxglove				Supplejack		
<i>Agalinis skinneriana</i>		E	—	<i>Botrychium matricariifolium</i>	E	—
Pale false foxglove				Matricary grapefern		
<i>Agastache scrophulariifolia</i>		S	—	<i>Botrychium oneidense</i>	E	—
Purple giant hyssop				Blunt-lobed grapefern		

Table 1. Continued.

	Status			Status	
	KSNPC	US		KSNPC	US
<i>Bouteloua curtipendula</i>	S	—	<i>Carex seorsa</i>	S	—
Side-oats grama			Weak stellate sedge		
<i>Boykinia aconitifolia</i>	T	—	<i>Carex stipata</i> var. <i>maxima</i>	S	—
Brook saxifrage			Stalkgrain sedge		
<i>Cabomba caroliniana</i>	T	—	<i>Carex straminea</i>	T	—
Carolina fanwort			Straw sedge		
<i>Calamagrostis canadensis</i> var.	E	—	<i>Carex tetanica</i>	E	—
<i>macouniana</i>			Rigid sedge		
Blue-joint reed grass			<i>Carya aquatica</i>	T	—
<i>Calamagrostis porteri</i> ssp. <i>insperata</i>	E	—	Water hickory		
Reed bent grass			<i>Castanea dentata</i>	E	—
<i>Calamagrostis porteri</i> ssp. <i>porteri</i>	T	—	American chestnut		
Porter's reed grass			<i>Castanea pumila</i>	T	—
<i>Callirhoe alcaeoides</i>	H	—	Allegheny chinkapin		
Clustered poppy-mallow			<i>Castilleja coccinea</i>	E	—
<i>Calopogon tuberosus</i>	E	—	Scarlet indian paintbrush		
Grass-pink			<i>Ceanothus herbaceus</i>	T	—
<i>Calycanthus floridus</i> var. <i>glaucus</i>	T	—	Prairie redroot		
Sweetshrub			<i>Cheilanthes alabamensis</i>	E	—
<i>Calylophus serrulatus</i>	H	—	Alabama lip fern		
Yellow evening primrose			<i>Cheilanthes feei</i>	E	—
<i>Carex aestivalis</i>	E	—	Fée's lip fern		
Summer sedge			<i>Chelone obliqua</i> var. <i>obliqua</i>	E	—
<i>Carex alata</i>	T	—	Red turtlehead		
Broadwing sedge			<i>Chelone obliqua</i> var. <i>speciosa</i>	S	—
<i>Carex appalachica</i>	T	—	Rose turtlehead		
Appalachian sedge			<i>Chrysogonum virginianum</i>	E	—
<i>Carex atlantica</i> ssp. <i>capillacea</i>	E	—	Green-and-gold		
Prickly bog sedge			<i>Chrysosplenium americanum</i>	E	—
<i>Carex austrocaroliniana</i>	S	—	American golden-saxifrage		
Tarheel sedge			<i>Cimicifuga rubifolia</i>	T	—
<i>Carex buxbaumii</i>	H	—	Appalachian bugbane		
Brown bog sedge			<i>Circaea alpina</i>	S	—
<i>Carex comosa</i>	H	—	Small enchanter's-nightshade		
Bristly sedge			<i>Clematis crispa</i>	T	—
<i>Carex crawei</i>	S	—	Blue jasmine leather-flower		
Crawe's sedge			<i>Coeloglossum viride</i> var. <i>virescens</i>	H	—
<i>Carex crebriflora</i>	T	—	Long-bract green orchis		
Coastal Plain sedge			<i>Collinsonia verticillata</i>	E	—
<i>Carex decomposita</i>	T	—	Whorled horse-balm		
Epiphytic sedge			<i>Comptonia peregrina</i>	E	—
<i>Carex gigantea</i>	T	—	Sweet-fern		
Large sedge			<i>Conradina verticillata</i>	E	T
<i>Carex hystericina</i>	H	—	Cumberland rosemary		
Porcupine sedge			<i>Convallaria montana</i>	E	—
<i>Carex joorii</i>	E	—	American lily-of-the-valley		
Cypress-swamp sedge			<i>Corallorhiza maculata</i>	E	—
<i>Carex juniperorum</i>	E	—	Spotted coralroot		
Cedar sedge			<i>Coreopsis pubescens</i>	S	—
<i>Carex lanuginosa</i>	E	—	Star tickseed		
Woolly sedge			<i>Corydalis sempervirens</i>	S	—
<i>Carex leptoneuria</i>	E	—	Pale corydalis		
Finely-nerved sedge			<i>Cymophyllus fraserianus</i>	E	—
<i>Carex reniformis</i>	E	—	Fraser's sedge		
Reniform sedge			<i>Cyperus plukenetii</i>	H	—
<i>Carex roanensis</i>	E	—	Plukenet's cyperus		
Roan sedge			<i>Cypripedium candidum</i>	E	—
<i>Carex rugosperma</i>	T	—	Small white lady's-slipper		
Umbel-like sedge			<i>Cypripedium kentuckiense</i>	S	—
			Kentucky lady's slipper		

Table 1. Continued.

	Status			Status	
	KSNPC	US		KSNPC	US
<i>Cypripedium parviflorum</i> Small yellow lady's-slipper	T	—	<i>Gentiana flavida</i> Yellow gentian	E	—
<i>Cypripedium reginae</i> Showy lady's-slipper	H	—	<i>Gentiana puberulenta</i> Prairie gentian	E	—
<i>Dalea purpurea</i> Purple prairie-clover	S	—	<i>Glandularia canadensis</i> Rose verbena	T	—
<i>Delphinium carolinianum</i> Carolina larkspur	T	—	<i>Gleditsia aquatica</i> Water locust	S	—
<i>Deschampsia cespitosa</i> ssp. <i>glauca</i> Tufted hair grass	E	—	<i>Glyceria acutiflora</i> Sharp-scaled manna grass	T	—
<i>Deschampsia flexuosa</i> Crinkled hair grass	T	—	<i>Gnaphalium helleri</i> var. <i>micradenium</i> Small rabbit-tobacco	H	—
<i>Dichanthelium boreale</i> Northern witch grass	S	—	<i>Gratiola pilosa</i> Shaggy hedge-hyssop	T	—
<i>Didiplis diandra</i> Water-purslane	S	—	<i>Gratiola viscidula</i> Short's hedge-hyssop	S	—
<i>Disporum maculatum</i> Nodding mandarin	S	—	<i>Gymnopogon ambiguus</i> Bearded skeleton grass	S	—
<i>Dodecatheon frenchii</i> French's shooting-star	S	—	<i>Gymnopogon brevifolius</i> Shortleaf skeleton grass	E	—
<i>Draba cuneifolia</i> Wedge-leaf whitlow-grass	E	—	<i>Halesia tetraptera</i> Common silverbell	T	—
<i>Drosera brevifolia</i> Dwarf sundew	E	—	<i>Hedeoma hispidum</i> Rough pennyroyal	T	—
<i>Drosera intermedia</i> Spoon-leaved sundew	H	—	<i>Helianthemum bicknellii</i> Plains frostweed	T	—
<i>Dryopteris carthusiana</i> Spinulose wood fern	S	—	<i>Helianthemum canadense</i> Canada frostweed	E	—
<i>Dryopteris ludoviciana</i> Southern shield wood fern	H	—	<i>Helianthus eggertii</i> Eggert's sunflower	T	T
<i>Echinodorus berteroi</i> Burhead	T	—	<i>Helianthus silphiodides</i> Silphium sunflower	E	—
<i>Echinodorus parvulus</i> Dwarf burhead	E	—	<i>Heracleum lanatum</i> Cow-parsnip	E	—
<i>Eleocharis olivacea</i> Olivaceous sedge	S	—	<i>Heteranthera dubia</i> Grassleaf mud-plantain	S	—
<i>Elodea nuttallii</i> Waterweed	T	—	<i>Heteranthera limosa</i> Blue mud-plantain	S	—
<i>Elymus svensonii</i> Svenson's wild rye	S	—	<i>Heterotheca subaxillaris</i> var. <i>latifolia</i> Broad-leaf golden-aster	T	—
<i>Eriophorum virginicum</i> Tawny cotton-grass	E	—	<i>Hexastylis contracta</i> Southern heartleaf	E	—
<i>Eryngium integrifolium</i> Blue-flower coyote-thistle	E	—	<i>Hexastylis heterophylla</i> Variable-leaved heartleaf	S	—
<i>Erythronium rostratum</i> Golden-star	S	—	<i>Hieracium longipilum</i> Hairy hawkweed	T	—
<i>Eupatorium maculatum</i> Spotted joe-pye-weed	H	—	<i>Houstonia serpyllifolia</i> Michaux's bluets	E	—
<i>Eupatorium semiserratum</i> Small-flowered thoroughwort	E	—	<i>Hydrocotyle americana</i> American water-pennywort	E	—
<i>Eupatorium steelei</i> Steele's joe-pye-weed	E	—	<i>Hydrolea ovata</i> Ovate fiddleleaf	E	—
<i>Euphorbia mercurialina</i> Mercury spurge	T	—	<i>Hydrolea uniflora</i> One-flower fiddleleaf	S	—
<i>Fimbristylis puberula</i> Hairy fimbristylis	T	—	<i>Hydrophyllum virginianum</i> Virginia waterleaf	S	—
<i>Forestiera ligustrina</i> Upland privet	T	—	<i>Hypericum adpressum</i> Creeping St. John's-wort	H	—
<i>Gentiana decora</i> Showy gentian	S	—	<i>Hypericum crux-andreae</i> St. Peter's-wort	T	—

Table 1. Continued.

	Status			Status	
	KSNPC	US		KSNPC	US
<i>Hypericum nudiflorum</i>	H	—	<i>Lobelia nuttallii</i>	T	—
Pretty St. John's-wort			Nuttall's lobelia		
<i>Hypericum pseudomaculatum</i>	H	—	<i>Lonicera dioica</i> var. <i>orientalis</i>	E	—
Large spotted St. John's-wort			Wild honeysuckle		
<i>Iris fulva</i>	E	—	<i>Lonicera reticulata</i>	E	—
Copper iris			Grape honeysuckle		
<i>Isoetes butleri</i>	E	—	<i>Ludwigia hirtella</i>	E	—
Butler's quillwort			Hairy ludwigia		
<i>Isoetes melanopoda</i>	E	—	<i>Lycopodiella appressa</i>	E	—
Blackfoot quillwort			Southern bog club-moss		
<i>Juglans cinerea</i>	S	—	<i>Lycopodiella inundata</i>	E	—
White walnut			Northern bog club-moss		
<i>Juncus articulatus</i>	S	—	<i>Lycopodium clavatum</i>	E	—
Jointed rush			Running-pine		
<i>Juncus elliotii</i>	H	—	<i>Lysimachia fraseri</i>	E	—
Bog rush			Fraser's loosestrife		
<i>Juncus filipendulus</i>	T	—	<i>Lysimachia radicans</i>	H	—
Long-styled rush			Trailing loosestrife		
<i>Juniperus communis</i> var. <i>depressa</i>	T	—	<i>Lysimachia terrestris</i>	E	—
Ground juniper			Swamp-candles		
<i>Koeleria macrantha</i>	E	—	<i>Maianthemum canadense</i>	T	—
June grass			Wild lily-of-the-valley		
<i>Krigia occidentalis</i>	E	—	<i>Maianthemum stellatum</i>	E	—
Western dwarf dandelion			Starry false solomon-seal		
<i>Lathyrus palustris</i>	T	—	<i>Malus angustifolia</i>	S	—
Vetchling peavine			Southern crabapple		
<i>Lathyrus venosus</i>	S	—	<i>Malvastrum hispidum</i>	T	—
Smooth veiny peavine			Hispid false mallow		
<i>Leavenworthia exigua</i> var. <i>laciniata</i>	T	—	<i>Marshallia grandiflora</i>	E	—
Glade cress			Barbara's-buttons		
<i>Leavenworthia torulosa</i>	T	—	<i>Matelea carolinensis</i>	E	—
Necklace glade cress			Carolina anglepod		
<i>Leiophyllum buxifolium</i>	H	—	<i>Melampyrum lineare</i> var. <i>latifolium</i>	T	—
Sand-myrtle			American cow-wheat		
<i>Lespedeza capitata</i>	S	—	<i>Melampyrum lineare</i> var. <i>pectinatum</i>	E	—
Round-head bush-clover			American crow-wheat		
<i>Lespedeza stuevei</i>	S	—	<i>Melanthera nivea</i>	S	—
Tall bush-clover			Snow melanthera		
<i>Lesquerella globosa</i>	T	C	<i>Melanthium parviflorum</i>	E	—
Lesquereux's bladderpod			Small-flowered false hellebore		
<i>Lesquerella lescurii</i>	S	—	<i>Melanthium virginicum</i>	E	—
Lescur's bladderpod			Virginia bunchflower		
<i>Leucothoe recurva</i>	E	—	<i>Melanthium woodii</i>	T	—
Fetterbush			False hellebore		
<i>Liatris cylindracea</i>	T	—	<i>Minuartia cumberlandensis</i>	E	E
Slender blazingstar			Cumberland sandwort		
<i>Lilium philadelphicum</i>	T	—	<i>Minuartia glabra</i>	T	—
Wood lily			Appalachian sandwort		
<i>Lilium superbum</i>	T	—	<i>Mirabilis albida</i>	E	—
Turk's cap lily			Pale umbrella-wort		
<i>Limnobium spongia</i>	T	—	<i>Monarda punctata</i>	H	—
American frog's-bit			Spotted beebalm		
<i>Liparis loeselii</i>	T	—	<i>Monotropsis odorata</i>	T	—
Loesel's twayblade			Sweet pinesap		
<i>Listera australis</i>	E	—	<i>Muhlenbergia bushii</i>	E	—
Southern twayblade			Bush's muhly		
<i>Listera smallii</i>	T	—	<i>Muhlenbergia cuspidata</i>	T	—
Kidney-leaf twayblade			Plains muhly		
<i>Lobelia appendiculata</i> var. <i>gattingeri</i>	E	—	<i>Muhlenbergia glabriflora</i>	S	—
Gattinger's lobelia			Hair grass		

Table 1. Continued.

	Status			Status	
	KSNPC	US		KSNPC	US
<i>Myriophyllum heterophyllum</i>	S	—	<i>Platanthera cristata</i>	T	—
Broadleaf water-milfoil			Yellow-crested orchid		
<i>Myriophyllum pinnatum</i>	H	—	<i>Platanthera integrilabia</i>	T	C
Cutleaf water-milfoil			White fringeless orchid		
<i>Najas gracillima</i>	S	—	<i>Platanthera psycodes</i>	E	—
Thread-like naiad			Small purple-fringed orchid		
<i>Nemophila aphylla</i>	T	—	<i>Poa saltuensis</i>	E	—
Small-flower baby-blue-eyes			Drooping blue grass		
<i>Nestronia umbellula</i>	E	—	<i>Podostemum ceratophyllum</i>	S	—
Conjurer's-nut			Threadfoot		
<i>Oenothera linifolia</i>	E	—	<i>Pogonia ophioglossoides</i>	E	—
Thread-leaf sundrops			Rose pogonia		
<i>Oenothera oakesiana</i>	H	—	<i>Polygala cruciata</i>	E	—
Evening primrose			Cross-leaf milkwort		
<i>Oenothera perennis</i>	E	—	<i>Polygala nuttallii</i>	H	—
Small sundrops			Nuttall's milkwort		
<i>Oenothera triloba</i>	T	—	<i>Polygala paucifolia</i>	E	—
Stemless evening-primrose			Gaywings		
<i>Oldenlandia uniflora</i>	E	—	<i>Polygala polygama</i>	T	—
Clustered bluets			Racemed milkwort		
<i>Onosmodium molle</i> ssp.	E	—	<i>Polymnia laevigata</i>	E	—
<i>hispidissimum</i>			Tennessee leafcup		
Hairy false gromwell			<i>Pontederia cordata</i>	T	—
<i>Onosmodium molle</i> ssp. <i>molle</i>	E	—	Pickernel-weed		
Soft false gromwell			<i>Potamogeton illinoensis</i>	S	—
<i>Onosmodium molle</i> ssp. <i>occidentale</i>	E	—	Illinois pondweed		
Western false gromwell			<i>Potamogeton pulcher</i>	T	—
<i>Orobanche ludoviciana</i>	H	—	Spotted pondweed		
Louisiana broomrape			<i>Prenanthes alba</i>	E	—
<i>Orontium aquaticum</i>	T	—	White rattlesnake-root		
Goldenclub			<i>Prenanthes aspera</i>	E	—
<i>Oxalis priceae</i>	H	—	Rough rattlesnake-root		
Price's yellow wood sorrel			<i>Prenanthes barbata</i>	E	—
<i>Parnassia asarifolia</i>	E	—	Barbed rattlesnake-root		
Kidney-leaf grass-of-parnassus			<i>Prenanthes crepidinea</i>	T	—
<i>Parnassia grandifolia</i>	E	—	Nodding rattlesnake-root		
Largeleaf grass-of-parnassus			<i>Psoralidium tenuiflorum</i>	E	—
<i>Paronychia argyrocoma</i>	E	—	Few-flowered scurf-pea		
Silvering			<i>Ptilimnium capillaceum</i>	T	—
<i>Paspalum boscianum</i>	S	—	Mock bishop's-weed		
Bull paspalum			<i>Ptilimnium costatum</i>	S	—
<i>Paxistima canbyi</i>	T	—	Eastern mock bishop's-weed		
Canby's mountain-lover			<i>Ptilimnium nuttallii</i>	E	—
<i>Pedicularis lanceolata</i>	H	—	Nuttall's mock bishop's-weed		
Swamp lousewort			<i>Pycnanthemum albescens</i>	E	—
<i>Perideridia americana</i>	T	—	White-leaved mountain-mint		
Eastern eulophus			<i>Pycnanthemum muticum</i>	T	—
<i>Phacelia ranunculacea</i>	S	—	Blunt mountain-mint		
Blue scorpion-weed			<i>Pyrola americana</i>	H	—
<i>Philadelphus inodorus</i>	T	—	American wintergreen		
Mock orange			<i>Ranunculus ambigens</i>	S	—
<i>Philadelphus pubescens</i>	E	—	Water-plantain spearwort		
Hoary mock orange			<i>Rhododendron canescens</i>	E	—
<i>Phlox bifida</i> ssp. <i>bifida</i>	T	—	Hoary azalea		
Cleft phlox			<i>Rhynchosia tomentosa</i>	E	—
<i>Phlox bifida</i> ssp. <i>stellaria</i>	T	—	Hairy snout-bean		
Starry cleft phlox			<i>Rhynchospora globularis</i>	S	—
<i>Plantago cordata</i>	H	—	Globe beaked-rush		
Heartleaf plantain			<i>Rhynchospora macrostachya</i>	E	—
			Tall beaked-rush		

Table 1. Continued.

	Status			Status	
	KSNPC	US		KSNPC	US
<i>Rubus canadensis</i>	E	—	<i>Silene regia</i>	E	—
Smooth blackberry			Royal catchfly		
<i>Rubus whartoniae</i>	T	—	<i>Silphium laciniatum</i> var. <i>laciniatum</i>	E	—
Wharton's dewberry			Compassplant		
<i>Rudbeckia subtomentosa</i>	E	—	<i>Silphium laciniatum</i> var. <i>robinsonii</i>	T	—
Sweet coneflower			Compassplant		
<i>Sabatia campanulata</i>	E	—	<i>Silphium pinnatifidum</i>	S	—
Slender marsh-pink			Tansy rosinweed		
<i>Sagittaria graminea</i>	T	—	<i>Silphium wasiotense</i>	S	—
Grass-leaf arrowhead			Appalachian rosinweed		
<i>Sagittaria platyphylla</i>	T	—	<i>Solidago albopilosa</i>	T	T
Delta arrowhead			White-haired goldenrod		
<i>Sagittaria rigida</i>	E	—	<i>Solidago buckleyi</i>	S	—
Sessile-fruit arrowhead			Buckley's goldenrod		
<i>Salix amygdaloides</i>	H	—	<i>Solidago curtisii</i>	T	—
Peachleaf willow			Curtis' goldenrod		
<i>Salix discolor</i>	H	—	<i>Solidago gracillima</i>	S	—
Pussy willow			Southern bog goldenrod		
<i>Salvia urticifolia</i>	E	—	<i>Solidago puberula</i>	S	—
Nettle-leaf sage			Downy goldenrod		
<i>Sambucus racemosa</i> ssp. <i>pubens</i>	E	—	<i>Solidago roanensis</i>	T	—
Red elderberry			Roan Mountain goldenrod		
<i>Sanguisorba canadensis</i>	E	—	<i>Solidago shortii</i>	E	E
Canada burnet			Short's goldenrod		
<i>Saxifraga michauxii</i>	T	—	<i>Solidago simplex</i> ssp. <i>randii</i>	S	—
Michaux's saxifrage			Rand's goldenrod		
<i>Saxifraga micranthidifolia</i>	E	—	<i>Solidago squarrosa</i>	H	—
Lettuce-leaf saxifrage			Squarrose goldenrod		
<i>Saxifraga pennsylvanica</i>	H	—	<i>Sparganium eurycarpum</i>	E	—
Swamp saxifrage			Large bur-reed		
<i>Schisandra glabra</i>	E	—	<i>Sphenopholis pennsylvanica</i>	S	—
Bay starvine			Swamp wedgescale		
<i>Schizachne purpurascens</i>	T	—	<i>Spiraea alba</i> var. <i>alba</i>	E	—
Purple-oat			Narrow-leaved meadowsweet		
<i>Schwalbea americana</i>	H	E	<i>Spiraea virginiana</i>	T	T
American chaffseed			Virginia spiraea		
<i>Scirpus expansus</i>	E	—	<i>Spiranthes lucida</i>	T	—
Woodland beak-rush			Shining ladies'-tresses		
<i>Scirpus fluviatilis</i>	E	—	<i>Spiranthes magnicamporum</i>	T	—
River bulrush			Great Plains ladies'-tresses		
<i>Scirpus hallii</i>	E	—	<i>Spiranthes ochroleuca</i>	S	—
Hall's bulrush			Yellow nodding ladies'-tresses		
<i>Scirpus heterochaetus</i>	E	—	<i>Spiranthes odorata</i>	E	—
Slender bulrush			Sweetscent ladies'-tresses		
<i>Scirpus microcarpus</i>	E	—	<i>Sporobolus clandestinus</i>	T	—
Small-fruit bulrush			Rough dropseed		
<i>Scirpus verecundus</i>	E	—	<i>Sporobolus heterolepis</i>	E	—
Bashful bulrush			Northern dropseed		
<i>Scleria ciliata</i> var. <i>ciliata</i>	E	—	<i>Stachys eplingii</i>	E	—
Fringed nut-rush			Epling's hedge-nettle		
<i>Scutellaria arguta</i>	T	—	<i>Stellaria fontinalis</i>	T	—
Hairy skullcap			Water stitchwort		
<i>Scutellaria saxatilis</i>	T	—	<i>Stellaria longifolia</i>	S	—
Rock skullcap			Longleaf stitchwort		
<i>Sedum telephioides</i>	T	—	<i>Streptopus roseus</i> var. <i>perspectus</i>	E	—
Allegheny stonecrop			Rosy twistedstalk		
<i>Sida hermaphrodita</i>	S	—	<i>Symphoricarpos albus</i>	E	—
Virginia-mallow			Snowberry		
<i>Silene ovata</i>	T	—	<i>Talinum calcaricum</i>	E	—
Ovate catchfly			Limestone fameflower		

Table 1. Continued.

	Status			Status	
	KSNPC	US		KSNPC	US
<i>Talinum teretifolium</i>	T	—	<i>Vitis labrusca</i>	S	—
Roundleaf fameflower			Northern fox grape		
<i>Taxus canadensis</i>	T	—	<i>Vitis rupestris</i>	T	—
Canadian yew			Sand grape		
<i>Tephrosia spicata</i>	E	—	<i>Woodsia appalachiana</i>	E	—
Spiked hoary-pea			Mountain woodsia		
<i>Thaspium pinnatifidum</i>	T	—	<i>Xerophyllum asphodeloides</i>	H	—
Cutleaf meadow-parsnip			Eastern turkeybeard		
<i>Thermopsis mollis</i>	E	—	<i>Xyris difformis</i>	E	—
Soft-haired thermopsis			Carolina yellow-eye-grass		
<i>Thuja occidentalis</i>	T	—	<i>Zizania palustris</i> var. <i>interior</i>	H	—
Northern white-cedar			Indian wild rice		
<i>Torreyochloa pallida</i>	E	—	<i>Zizaniopsis miliacea</i>	T	—
Pale manna grass			Southern wild rice		
<i>Toxicodendron vernix</i>	E	—	Animals		
Poison sumac			Gastropods		
<i>Tragia urticifolia</i>	E	—	<i>Anguispira rugoderma</i>	T	—
Nettle-leaf noseburn			Pine Mountain tigersnail		
<i>Trepocarpus aethusae</i>	T	—	<i>Antroselatus spiralis</i>	S	—
Trepocarpus			Shaggy cavesnail		
<i>Trichostema setaceum</i>	E	—	<i>Appalachina chilhoweensis</i>	S	—
Narrow-leaved bluecurls			Queen crater		
<i>Trientalis borealis</i>	E	—	<i>Fumonelix wetherbyi</i>	S	—
Northern starflower			Clifty covert		
<i>Trifolium reflexum</i>	E	—	<i>Glyphyalinia raderi</i>	S	—
Buffalo clover			Maryland glyph		
<i>Trifolium stoloniferum</i>	T	E	<i>Glyphyalinia rhoadsi</i>	T	—
Running buffalo clover			Sculpted glyph		
<i>Trillium nivale</i>	E	—	<i>Helicodiscus notius specus</i>	T	—
Snow trillium			A snail		
<i>Trillium pusillum</i> var. <i>ozarkanum</i>	E	—	<i>Helicodiscus punctatellus</i>	S	—
Ozark least trillium			Punctate coil		
<i>Trillium pusillum</i> var. <i>pusillum</i>	E	—	<i>Leptoxis praerosa</i>	S	—
Least trillium			Onyx rocksnail		
<i>Trillium undulatum</i>	T	—	<i>Lithasia armigera</i>	S	—
Painted trillium			Armored rocksnail		
<i>Triplasis purpurea</i>	H	—	<i>Lithasia geniculata</i>	S	—
Purple sand grass			Ornate rocksnail		
<i>Ulmus serotina</i>	S	—	<i>Lithasia salebroso</i>	S	—
September elm			Muddy rocksnail		
<i>Utricularia macrorrhiza</i>	E	—	<i>Lithasia verrucosa</i>	S	—
Greater bladderwort			Varicose rocksnail		
<i>Vallisneria americana</i>	S	—	<i>Mesomphix rugeli</i>	T	—
Eel-grass			Wrinkled button		
<i>Vernonia noveboracensis</i>	S	—	<i>Neohelix dentifera</i>	T	—
New York ironweed			Big-tooth whitelip		
<i>Veronica americana</i>	H	—	<i>Patera panselenus</i>	S	—
American speedwell			Virginia bladetooth		
<i>Viburnum molle</i>	T	—	<i>Pilsbryna</i> sp.	E	—
Missouri arrow-wood			A snail (undescribed)		
<i>Viburnum nudum</i>	E	—	<i>Pleurocera alveare</i>	S	—
Possum haw viburnum			Rugged hornsnail		
<i>Viburnum rafinesquianum</i> var. <i>rafinesquianum</i>	T	—	<i>Pleurocera curta</i>	S	—
Downy arrowwood			Shortspire hornsnail		
<i>Viola septemloba</i> var. <i>egglestonii</i>	S	—	<i>Rabdotus dealbatus</i>	T	—
Eggleston's violet			Whitewashed rabdotus		
<i>Viola walteri</i>	T	—	<i>Rhodacme elatior</i>	S	—
Walter's violet			Domed ancyliid		

Table 1. Continued.

	Status			Status	
	KSNPC	US		KSNPC	US
<i>Vertigo bollesiana</i>	E	—	<i>Potamilus purpuratus</i>	E	—
Delicate vertigo			Bleufer		
<i>Vertigo clappi</i>	E	—	<i>Ptychobranhus subtentum</i>	E	C
Cupped vertigo			Fluted kidneyshell		
<i>Vitrinizonites latissimus</i>	T	—	<i>Quadrula cylindrica cylindrica</i>	T	—
Glassy grapeskin			Rabbitsfoot		
<i>Webbhelix multilineata</i>	T	—	<i>Simpsonaias ambigua</i>	T	—
Striped whitelip			Salamander mussel		
Freshwater Mussels			<i>Toxolasma lividus</i>	E	—
<i>Alasmidonta atropurpurea</i>	E	E	Purple lilliput		
Cumberland elktoe			<i>Toxolasma texasiensis</i>	E	—
<i>Alasmidonta marginata</i>	T	—	Texas lilliput		
Elktoe			<i>Villosa fabalis</i>	E	—
<i>Anodontoides denigratus</i>	E	—	Rayed bean		
Cumberland papershell			<i>Villosa lienosa</i>	S	—
<i>Cumberlandia monodonta</i>	E	—	Little spectaclecase		
Spectaclecase			<i>Villosa ortmanni</i>	T	—
<i>Cyprogenia stegaria</i>	E	E	Kentucky creekshell		
Fanshell			<i>Villosa trabalis</i>	E	E
<i>Epioblasma brevidens</i>	E	E	Cumberland bean		
Cumberlandian combshell			<i>Villosa vanuxemensis</i>	T	—
<i>Epioblasma capsaeformis</i>	E	E	Mountain creekshell		
Oyster mussel			Crustaceans		
<i>Epioblasma obliquata obliquata</i>	E	E	<i>Barbicambarus cornutus</i>	S	—
Catspaw			Bottlebrush crayfish		
<i>Epioblasma torulosa rangiana</i>	E	E	<i>Bryocamptus morrisoni elegans</i>	T	—
Northern riffleshell			A copepod		
<i>Epioblasma triquetra</i>	S	—	<i>Caacidotea barri</i>	E	—
Snuffbox			Clifton cave isopod		
<i>Fusconaia subrotunda subrotunda</i>	S	—	<i>Cambarellus puer</i>	E	—
Longsolid			A dwarf crayfish		
<i>Lampsilis abrupta</i>	E	E	<i>Cambarellus shufeldtii</i>	S	—
Pink mucket			Cajun dwarf crayfish		
<i>Lampsilis ovata</i>	E	—	<i>Cambarus parvoculus</i>	E	—
Pocketbook			A crayfish		
<i>Lasmigona compressa</i>	E	—	<i>Cambarus veteranus</i>	S	—
Creek heelsplitter			A crayfish		
<i>Lasmigona subviridis</i>	E	—	<i>Gammarus bousfieldi</i>	E	—
Green floater			Bousfield's amphipod		
<i>Lexingtonia dolabelloides</i>	H	C	<i>Macrobrachium ohione</i>	E	—
Slabside pearlymussel			Ohio shrimp		
<i>Obovaria retusa</i>	E	E	<i>Orconectes australis</i>	T	—
Ring pink			A crayfish		
<i>Pegias fabula</i>	E	E	<i>Orconectes bisectus</i>	T	—
Littlewing pearlymussel			Crittenden crayfish		
<i>Plethobasus cooperianus</i>	E	E	<i>Orconectes burri</i>	T	—
Orangefoot pimpleback			A crayfish		
<i>Plethobasus cyphus</i>	S	—	<i>Orconectes inermis</i>	S	—
Sheepnose			A crayfish		
<i>Pleurobema clava</i>	E	E	<i>Orconectes jeffersoni</i>	E	—
Clubshell			Louisville crayfish		
<i>Pleurobema oviforme</i>	E	—	<i>Orconectes lancifer</i>	E	—
Tennessee clubshell			A crayfish		
<i>Pleurobema plenum</i>	E	E	<i>Orconectes palmeri</i>	E	—
Rough pigtoe			A crayfish		
<i>Pleurobema rubrum</i>	E	—	<i>Orconectes pellucidus</i>	S	—
Pyramid pigtoe			A crayfish		
<i>Potamilus capax</i>	E	E	<i>Palaemonias ganteri</i>	E	E
Fat pocketbook			Mammoth Cave shrimp		

Table 1. Continued.

	Status			Status	
	KSNPC	US		KSNPC	US
<i>Procambarus viaeviridis</i>	T	—	<i>Pseudanophthalmus horni abditus</i>	T	—
A crayfish			Concealed cave beetle		
<i>Stygobromus vitreus</i>	S	—	<i>Pseudanophthalmus horni caecus</i>	T	—
An amphipod			Clifton Cave beetle		
Insects			<i>Pseudanophthalmus horni horni</i>	S	—
<i>Calephelis mutica</i>	S	—	Garman's cave beetle		
Swamp metalmark			<i>Pseudanophthalmus hypolithos</i>	T	—
<i>Callophrys irus</i>	S	—	Ashcamp cave beetle		
Frosted elfin			<i>Pseudanophthalmus inexpectatus</i>	T	—
<i>Celithemis verna</i>	S	—	Surprising cave beetle		
Double-ringed pennant			<i>Pseudanophthalmus parvus</i>	T	—
<i>Cheumatopsyche helma</i>	H	—	Tatum Cave beetle		
Helma's net-spinning caddisfly			<i>Pseudanophthalmus pholeter</i>	E	—
<i>Dryobius sexnotatus</i>	T	—	Greater Adams Cave beetle		
Sixbanded longhorn beetle			<i>Pseudanophthalmus pubescens</i>	T	—
<i>Ephemerella inconstans</i>	H	—	<i>intrepidus</i>		
An ephemerellid mayfly			A cave beetle		
<i>Erora laeta</i>	S	—	<i>Pseudanophthalmus puteanus</i>	T	—
Early hairstreak			Old Well Cave beetle		
<i>Euphyes dukesi</i>	S	—	<i>Pseudanophthalmus rogersae</i>	T	—
Duke's skipper			Rogers' cave beetle		
<i>Litobranchia recurvata</i>	S	—	<i>Pseudanophthalmus scholasticus</i>	T	—
A burrowing mayfly			Scholarly cave beetle		
<i>Lordithon niger</i>	H	—	<i>Pseudanophthalmus simulans</i>	T	—
Black lordithon rove beetle			Cub Run Cave beetle		
<i>Lyttosis permagnaria</i>	E	—	<i>Pseudanophthalmus tenebrosus</i>	T	—
A geometrid moth			Stevens Creek Cave beetle		
<i>Manophylax butleri</i>	S	—	<i>Pseudanophthalmus troglodytes</i>	T	—
A limnephilid caddisfly			Louisville cave beetle		
<i>Nicrophorus americanus</i>	H	E	<i>Pyrgus wyandot</i>	T	—
American burying beetle			Appalachian grizzled skipper		
<i>Ophiogomphus aspersus</i>	H	—	<i>Raptoheptagenia cruentata</i>	H	—
Brook snaketail			A heptageniid mayfly		
<i>Ophiogomphus howei</i>	S	—	<i>Satyrium favonius ontario</i>	S	—
Pygmy snaketail			Northern hairstreak		
<i>Papaipema eryngii</i>	E	—	<i>Speyeria idalia</i>	H	—
Rattlesnake-master borer moth			Regal fritillary		
<i>Phyciodes batesii</i>	H	—	<i>Stenonema bednariki</i>	S	—
Tawny crescent			A heptageniid mayfly		
<i>Polygonia faunus</i>	H	—	<i>Stylurus notatus</i>	H	—
Green comma			Elusive clubtail		
<i>Polygonia progne</i>	H	—	<i>Traverella lewisi</i>	H	—
Gray comma			A leptophlebiid mayfly		
<i>Pseudanophthalmus audax</i>	T	—	Fishes		
Bold cave beetle			<i>Acipenser fulvescens</i>	E	—
<i>Pseudanophthalmus calcareus</i>	T	—	Lake sturgeon		
Limestone cave beetle			<i>Alosa alabamae</i>	E	C
<i>Pseudanophthalmus catoryctos</i>	E	—	Alabama shad		
Lesser Adams cave beetle			<i>Amblyopsis spelaea</i>	S	—
<i>Pseudanophthalmus conditus</i>	T	—	Northern cavefish		
Hidden cave beetle			<i>Ammocrypta clara</i>	E	—
<i>Pseudanophthalmus desertus major</i>	T	—	Western sand darter		
Beaver cave beetle			<i>Atractosteus spatula</i>	E	—
<i>Pseudanophthalmus exoticus</i>	H	—	Alligator gar		
Exotic cave beetle			<i>Cyprinella camura</i>	E	—
<i>Pseudanophthalmus frigidus</i>	T	—	Bluntnose shiner		
Icebox cave beetle			<i>Cyprinella venusta</i>	S	—
<i>Pseudanophthalmus globiceps</i>	T	—	Blacktail shiner		
Round-headed cave beetle					

Table 1. Continued.

	Status			Status	
	KSNPC	US		KSNPC	US
<i>Erimystax insignis</i>	E	—	<i>Macrhybopsis gelida</i>	H	C
Blotched chub			Sturgeon chub		
<i>Erimyzon sucetta</i>	T	—	<i>Macrhybopsis meeki</i>	H	C
Lake chubsucker			Sicklefin chub		
<i>Esox niger</i>	S	—	<i>Menidia beryllina</i>	T	—
Chain pickerel			Inland silverside		
<i>Etheostoma chienense</i>	E	E	<i>Moxostoma poecilurum</i>	E	—
Relict darter			Blacktail redhorse		
<i>Etheostoma cinereum</i>	S	—	<i>Nocomis biguttatus</i>	S	—
Ashy darter			Hornyhead chub		
<i>Etheostoma fusiforme</i>	E	—	<i>Notropis albizonatus</i>	E	E
Swamp darter			Palezone shiner		
<i>Etheostoma lynceum</i>	E	—	<i>Notropis hudsonius</i>	S	—
Brighteye darter			Spottail shiner		
<i>Etheostoma maculatum</i>	T	—	<i>Notropis maculatus</i>	T	—
Spotted darter			Taillight shiner		
<i>Etheostoma microlepidum</i>	E	—	<i>Notropis</i> sp.	E	—
Smallscale darter			Sawfin shiner (undescribed)		
<i>Etheostoma nigrum susanae</i>	E	C	<i>Noturus exilis</i>	E	—
Johnny darter			Slender madtom		
<i>Etheostoma parvipinne</i>	E	—	<i>Noturus hildebrandi</i>	E	—
Goldstripe darter			Least madtom		
<i>Etheostoma percnurum</i>	E	E	<i>Noturus phaeus</i>	E	—
Duskytail darter			Brown madtom		
<i>Etheostoma proeliare</i>	T	—	<i>Noturus stigmosus</i>	S	—
Cypress darter			Northern madtom		
<i>Etheostoma pyrrhogaster</i>	E	—	<i>Percina macrocephala</i>	T	—
Firebelly darter			Longhead darter		
<i>Etheostoma swaini</i>	E	—	<i>Percina squamata</i>	E	—
Gulf darter			Olive darter		
<i>Etheostoma tecumsehi</i>	T	—	<i>Percopsis omiscomaycus</i>	S	—
Shawnee darter			Trout-perch		
<i>Fundulus chrysotus</i>	E	—	<i>Phenacobius uranops</i>	S	—
Golden topminnow			Stargazing minnow		
<i>Fundulus dispar</i>	E	—	<i>Phoxinus cumberlandensis</i>	T	T
Northern starhead topminnow			Blackside dace		
<i>Hybognathus hayi</i>	E	—	<i>Platygobio gracilis</i>	S	—
Cypress minnow			Flathead chub		
<i>Hybognathus placitus</i>	S	—	<i>Rhinichthys cataractae</i>	E	—
Plains minnow			Longnose dace		
<i>Hybopsis amnis</i>	H	—	<i>Scaphirhynchus albus</i>	E	E
Pallid shiner			Pallid sturgeon		
<i>Ichthyomyzon castaneus</i>	S	—	<i>Thoburnia atripinnis</i>	S	—
Chestnut lamprey			Blackfin sucker		
<i>Ichthyomyzon fossor</i>	T	—	<i>Typhlichthys subterraneus</i>	S	—
Northern brook lamprey			Southern cavefish		
<i>Ichthyomyzon gagei</i>	H	—	<i>Umbra limi</i>	T	—
Southern brook lamprey			Central mudminnow		
<i>Ichthyomyzon greeleyi</i>	T	—	Amphibians		
Mountain brook lamprey			<i>Amphiuma tridactylum</i>	E	—
<i>Ictiobus niger</i>	S	—	Three-toed amphiuma		
Black buffalo			<i>Cryptobranchus alleganiensis</i>	S	—
<i>Lampetra appendix</i>	T	—	<i>alleganiensis</i>		
American brook lamprey			Eastern hellbender		
<i>Lepomis marginatus</i>	E	—	<i>Eurycea guttolineata</i>	T	—
Dollar sunfish			Three-lined salamander		
<i>Lepomis miniatus</i>	T	—	<i>Hyla avivoca</i>	T	—
Redspotted sunfish			Bird-voiced treefrog		
<i>Lota lota</i>	S	—			
Burbot					

Table 1. Continued.

	Status			Status	
	KSNPC	US		KSNPC	US
<i>Hyla cinerea</i>	S	—	<i>Ammodramus henslowii</i>	S	—
Green treefrog			Henslow's sparrow		
<i>Hyla gratiosa</i>	S	—	<i>Anas clypeata</i>	E	—
Barking treefrog			Northern shoveler		
<i>Hyla versicolor</i>	S	—	<i>Anas discors</i>	T	—
Gray treefrog			Blue-winged teal		
<i>Plethodon cinereus</i>	S	—	<i>Ardea alba</i>	E	—
Redback salamander			Great egret		
<i>Plethodon wehrlei</i>	E	—	<i>Ardea herodias</i>	S	—
Wehrle's salamander			Great blue heron		
<i>Rana areolata circulosa</i>	S	—	<i>Asio flammeus</i>	E	—
Northern crawfish frog			Short-eared owl		
<i>Rana pipiens</i>	S	—	<i>Asio otus</i>	E	—
Northern leopard frog			Long-eared owl		
Reptiles			<i>Bartramia longicauda</i>	H	—
<i>Apalone mutica mutica</i>	S	—	Upland sandpiper		
Midland smooth softshell			<i>Botaurus lentiginosus</i>	H	—
<i>Chrysemys picta dorsalis</i>	T	—	American bittern		
Southern painted turtle			<i>Bubulcus ibis</i>	S	—
<i>Clonophis kirtlandii</i>	T	—	Cattle egret		
Kirtland's snake			<i>Certhia americana</i>	E	—
<i>Elaphe guttata guttata</i>	S	—	Brown creeper		
Corn snake			<i>Chondestes grammacus</i>	T	—
<i>Eumeces anthracinus anthracinus</i>	T	—	Lark sparrow		
Northern coal skink			<i>Circus cyaneus</i>	T	—
<i>Eumeces anthracinus pluvialis</i>	E	—	Northern harrier		
Southern coal skink			<i>Cistothorus platensis</i>	S	—
<i>Eumeces inexpectatus</i>	S	—	Sedge wren		
Southeastern five-lined skink			<i>Corvus corax</i>	E	—
<i>Farancia abacura reinwardtii</i>	S	—	Common raven		
Western mud snake			<i>Corvus ossifragus</i>	S	—
<i>Lampropeltis triangulum elapsoides</i>	S	—	Fish crow		
Scarlet kingsnake			<i>Dendroica fusca</i>	T	—
<i>Macrolemys temminckii</i>	T	—	Blackburnian warbler		
Alligator snapping turtle			<i>Dolichonyx oryzivorus</i>	S	—
<i>Nerodia cyclopion</i>	E	—	Bobolink		
Mississippi green water snake			<i>Egretta caerulea</i>	E	—
<i>Nerodia erythrogaster neglecta</i>	S	—	Little blue heron		
Copperbelly water snake			<i>Empidonax minimus</i>	E	—
<i>Nerodia fasciata confluens</i>	E	—	Least flycatcher		
Broad-banded water snake			<i>Falco peregrinus</i>	E	—
<i>Ophisaurus attenuatus longicaudus</i>	T	—	Peregrine falcon		
Eastern slender glass lizard			<i>Fulica americana</i>	H	—
<i>Pituophis melanoleucus melanoleucus</i>	T	—	American coot		
Northern pine snake			<i>Gallinula chloropus</i>	T	—
<i>Sistrurus miliarius streckeri</i>	T	—	Common moorhen		
Western pigmy rattlesnake			<i>Haliaeetus leucocephalus</i>	E	T
<i>Thamnophis proximus proximus</i>	T	—	Bald eagle		
Western ribbon snake			<i>Ictinia mississippiensis</i>	S	—
<i>Thamnophis sauritus sauritus</i>	S	—	Mississippi kite		
Eastern ribbon snake			<i>Ixobrychus exilis</i>	T	—
Breeding Birds			Least bittern		
<i>Accipiter striatus</i>	S	—	<i>Junco hyemalis</i>	S	—
Sharp-shinned hawk			Dark-eyed junco		
<i>Actitis macularia</i>	E	—	<i>Lophodytes cucullatus</i>	T	—
Spotted sandpiper			Hooded merganser		
<i>Aimophila aestivalis</i>	E	—	<i>Nyctanassa violacea</i>	T	—
Bachman's sparrow			Yellow-crowned night-heron		
			<i>Nycticorax nycticorax</i>	T	—
			Black-crowned night-heron		

Table 1. Continued.

	Status			Status	
	KSNPC	US		KSNPC	US
<i>Pandion haliaetus</i>	T	—	Mammals		
Osprey			<i>Clethrionomys gapperi maurus</i>	S	—
<i>Passerculus sandwichensis</i>	S	—	Kentucky red-backed vole		
Savannah sparrow			<i>Corynorhinus rafinesquii</i>	S	—
<i>Phalacrocorax auritus</i>	H	—	Rafinesque's big-eared bat		
Double-crested cormorant			<i>Corynorhinus townsendii virginianus</i>	E	E
<i>Pheucticus ludovicianus</i>	S	—	Virginia big-eared bat		
Rose-breasted grosbeak			<i>Mustela nivalis</i>	S	—
<i>Picoides borealis</i>	E	E	Least weasel		
Red-cockaded woodpecker			<i>Myotis austroriparius</i>	E	—
<i>Podilymbus podiceps</i>	E	—	Southeastern myotis		
Pied-billed grebe			<i>Myotis grisescens</i>	E	E
<i>Poocetes gramineus</i>	E	—	Gray myotis		
Vesper sparrow			<i>Myotis leibii</i>	T	—
<i>Rallus elegans</i>	E	—	Eastern small-footed myotis		
King rail			<i>Myotis sodalis</i>	E	E
<i>Riparia riparia</i>	S	—	Indiana myotis		
Bank swallow			<i>Nycticeius humeralis</i>	T	—
<i>Sitta canadensis</i>	E	—	Evening bat		
Red-breasted nuthatch			<i>Peromyscus gossypinus</i>	T	—
<i>Sterna antillarum</i>	E	E	Cotton mouse		
Least tern			<i>Sorex cinereus</i>	S	—
<i>Thryomanes bewickii</i>	S	—	Masked shrew		
Bewick's wren			<i>Sorex dispar blitchi</i>	E	—
<i>Tyto alba</i>	S	—	Long-tailed shrew		
Barn owl			<i>Spilogale putorius</i>	S	—
<i>Vermivora chrysoptera</i>	T	—	Eastern spotted skunk		
Golden-winged warbler			<i>Ursus americanus</i>	S	—
<i>Vireo bellii</i>	S	—	Black bear		
Bell's vireo					
<i>Wilsonia canadensis</i>	S	—			
Canada warbler					

current status of Kentucky's rare organisms. Information about delisted and other taxa are maintained in manual files for use in the event that changes in distribution or status occur.

Each edition of these lists (Branson et al. 1981; Warren et al. 1986; KSNPC 1996, 1997, 1999) has been refined and enhanced with status changes and the addition of new taxonomic groups. As previously noted, these lists are important conservation tools used by KSNPC to focus protection efforts. We hope this information is used by planners and decision makers to conserve Kentucky's unique natural heritage through research, protection, and avoidance.

ACKNOWLEDGMENTS

We gratefully acknowledge the following contributors to this effort: Danny Barrett, U.S. Army Corps of Engineers; Steve R. Bloemer,

Tennessee Valley Authority; Hal D. Bryan, Eco-Tech, Inc.; Brooks M. Burr and Jeff G. Stewart, Southern Illinois University at Carbondale; Samuel M. Call and Michael C. Compton, Kentucky Division of Water; Julian J.N. Campbell, Kentucky Chapter of TNC; Ross C. Clark, Ronald L. Jones, Guenter A. Schuster, and Matthew R. Thomas, Eastern Kentucky University; H. Carl Cook, Florida State Collection of Arthropods; Charles V. Covell, Jr., University of Louisville; David J. Eisenhour, Les E. Meade, and Allen C. Risk, Morehead State University; Loran D. Gibson; James D. Kiser; Lewis E. Kornman, Kerry W. Prather, Douglas E. Stephens, Steven C. Thomas, and Traci A. Wethington, Kentucky Department of Fish and Wildlife Resources; James B. Layzer, Tennessee Technological University; John R. MacGregor and David D. Taylor, United States Forest Service; Christine

Table 2. Diversity and conservation status of the major groups of organisms in Kentucky, 2000.

Number of Kentucky species or taxa ¹	Lichens	Mosses	Vascular plants	Gastropods	Freshwater mussels	Crustaceans	Insects	Fishes	Amphibians	Reptiles	Breeding birds	Mammals
Native	unk.	326	2262	~ 259	103	unk.	unk.	237	51	52	168	70
Exotic	unk.	0	745	~ 12	0	unk.	unk.	23	0	1	4	5
KSNPC Monitored as Rare	1	17	372	25	36	20	48	61	11	18	49	14
KSNPC Endangered	1	12	151	3	26	9	4	28	2	3	18	5
KSNPC Threatened	0	5	116	8	5	5	20	13	2	8	11	3
KSNPC Special Concern	0	0	67	14	4	6	11	16	7	7	16	6
KSNPC Historical	0	0	38	0	1	0	13	4	0	0	4	0
Presumed Extinct or Extirpated	0	0	4	0	19	0	1	8	0	1	8	5
Extant/Extirpated												
Federally Endangered or Threatened	0/0	0/0	10/0	0/0	14/8	1/0	1/0	5/0	0/0	0/0	3/2	3/3
Extant/Extirpated												
Federally Proposed Endangered or Threatened	0/0	0/0	0/0	0/0	0/1	0/0	0/0	0/0	0/0	0/0	0/0	0/0
Extant Federal Candidate	0	0	2	0	2	0	0	4	0	0	0	0

¹ Totals include some distinctive subspecies and varieties for gastropods, freshwater mussels, and fishes but only species for lichens, mosses, vascular plants, amphibians, reptiles, breeding birds, and mammals. Totals do not include species recorded from Kentucky based on fossil specimens. Sources for totals of native taxa/species: mosses (Risk 1999; Snider et al. 1988); vascular plants (Medley 1993); gastropods, freshwater mussels, fishes, amphibians, reptiles, breeding birds, and mammals (KSNPC 2000).

Table 3. Plants and animals presumed extinct or extirpated from Kentucky.

	US Status		US Status
Plants		Fishes	
<i>Caltha palustris</i> var. <i>palustris</i>		<i>Ammocrypta vivax</i>	
Marsh marigold	—	Scaly sand darter	—
<i>Orbexilum stipulatum</i>		<i>Crystallaria asprella</i>	
Stipuled scurf-pea	—	Crystal darter	—
<i>Physostegia intermedia</i>		<i>Erimystax x-punctatus</i>	
Slender dragon-head	—	Gravel chub	—
<i>Polytaenia nuttallii</i>		<i>Etheostoma microperca</i>	
Prairie parsley	—	Least darter	—
Animals		<i>Hemitremia flammea</i>	
Freshwater Mussels		Flame chub	—
<i>Dromus dromas</i>		<i>Moxostoma lacerum</i>	
Dromedary pearlymussel	E	Harelip sucker	—
<i>Epioblasma arcaeformis</i>		<i>Moxostoma valenciennesi</i>	
Sugarspoon	—	Greater redhorse	—
<i>Epioblasma biemarginata</i>		<i>Percina burtoni</i>	
Angled riffleshell	—	Blotchside logperch	—
<i>Epioblasma flexuosa</i>		Reptiles	
Leafshell	—	<i>Masticophis flagellum flagellum</i>	
<i>Epioblasma florentina florentina</i>		Eastern coachwhip	—
Yellow blossom	E	Breeding Birds	
<i>Epioblasma florentina walkeri</i>		<i>Anhinga anhinga</i>	
Tan riffleshell	E	Anhinga	—
<i>Epioblasma haysiana</i>		<i>Campephilus principalis</i>	
Acornshell	—	Ivory-billed woodpecker	E
<i>Epioblasma lewisii</i>		<i>Chlidonias niger</i>	
Forkshell	—	Black tern	—
<i>Epioblasma obliquata perobliqua</i>		<i>Conuropsis carolinensis</i>	
White catspaw	E	Carolina parakeet	—
<i>Epioblasma personata</i>		<i>Ectopistes migratorius</i>	
Round combshell	—	Passenger pigeon	—
<i>Epioblasma propinqua</i>		<i>Elanoides forficatus forficatus</i>	
Tennessee riffleshell	—	Swallow-tailed kite	—
<i>Epioblasma sampsonii</i>		<i>Tympanuchus cupido</i>	
Wabash riffleshell	—	Greater prairie-chicken	—
<i>Epioblasma stewardsonii</i>		<i>Vermivora bachmanii</i>	
Cumberland leafshell	—	Bachman's warbler	E
<i>Epioblasma torulosa torulosa</i>		Mammals	
Tubercled blossom	E	<i>Bos bison</i>	
<i>Hemistena lata</i>		American bison	—
Cracking pearlymussel	E	<i>Canis lupus</i>	
<i>Leptodea leptodon</i>		Gray wolf	E
Scaleshell	PE	<i>Canis rufus</i>	
<i>Plethobasus cicatricosus</i>		Red wolf	E
White wartyback	E	<i>Cervus elaphus</i>	
<i>Quadrula fragosa</i>		Elk	—
Winged mapleleaf	E	<i>Puma concolor cougar</i>	
<i>Quadrula tuberosa</i>		Eastern puma	E
Rough rockshell	—		
Insects			
<i>Pentagenia robusta</i>			
Robust pentagenian burrowing mayfly	—		

A. Mayer and Christopher A. Taylor, Illinois Natural History Survey; Robert F.C. Naczi and John W. Thieret, Northern Kentucky University; Ralph Thompson, Berea College; and Charles E. Wright, Kentucky Department for Surface Mining Reclamation and Enforcement.

LITERATURE CITED

- Anderson, L. E. 1990. A checklist of *Sphagnum* in North America north of Mexico. *Bryologist* 93:500–501.
- [AOU] American Ornithologists' Union. 1998. Check-list of North American birds, 7th ed. American Ornithologists' Union, Washington, DC.
- Arnett, R. H., Jr. (ed). 1983. Checklist of the beetles of North and Central America and the West Indies. Flora and Fauna Publications, Gainesville, FL.
- Barr, T. C., Jr. 1968. Ecological studies in the Mammoth Cave system of Kentucky. I. The biota. *Int. J. Speleol.* 3:147–204.
- Barr, T. C., Jr. 1996. Cave beetle status survey and pre-listing recovery project. Report prepared for the U.S. Fish and Wildlife Service and Kentucky Department of Fish and Wildlife Resources, Frankfort, KY.
- Branson, B. A., D. F. Harker, Jr., J. M. Baskin, M. E. Medley, D. L. Batch, M. L. Warren, Jr., W. H. Davis, W. C. Houtcooper, B. L. Monroe, Jr., L. R. Phillippe, and P. Cupp. 1981. Endangered, threatened, and rare animals and plants of Kentucky. *Trans. Kentucky Acad. Sci.* 42:77–89.
- Cassie, B., J. Glassberg, P. A. Opler, R. K. Robbins, and G. Tudor. 1995. North American Butterfly Association (NABA) checklist and English names of North American butterflies. North American Butterfly Association, Morristown, NJ.
- Ceas, P. A., and L. M. Page. 1997. Systematic studies of the *Etheostoma spectabile* complex (Percidae; subgenus *Oligocephalus*), with descriptions of four new species. *Copeia* 1997:496–522.
- Collins, J. T. (ed). 1990. Standard common and current scientific names for North American amphibians and reptiles, 3rd ed. Society for the Study of Amphibians and Reptiles Herpetol. Circ. 19.
- Crum, L. A., H. A. Anderson, and W. R. Buck. 1990. List of mosses of North America north of Mexico. *Bryologist* 93:448–499.
- Egan, R. S. 1987. A fifth checklist of the lichen-forming and allied fungi of the continental United States and Canada. *Bryologist* 90:77–173.
- Frost, D. R. (ed). 1985. Amphibian species of the world: a taxonomic and geographical reference. Allen Press and the Association of Systematics Collections, Lawrence, KS.
- Gordon, M. E. 1995. *Anodontoides denigratus* (Lea) (Bivalvia: Unionoida) from the upper Cumberland River system. Report submitted to the U.S. Fish and Wildlife Service, Asheville, NC.
- Hall, E. R. 1981. The mammals of North America, 2nd ed. John Wiley and Sons, New York, NY.
- Holsinger, J. R. 1972. The freshwater amphipod crustaceans (Gammaridae) of North America. Water Pollution Control Research Series 18050 ELD04/72. U.S. Environmental Protection Agency, Cincinnati, OH.
- Hubricht, L. 1985. The distributions of the native land mollusks of the eastern United States. *Fieldiana: Zool.* 24:1–191.
- Jones, J. K., Jr., R. S. Hoffmann, D. W. Rice, C. Jones, R. J. Baker, and M. D. Engstrom. 1992. Revised checklist of North American mammals north of Mexico, 1991. *Occas. Papers Mus. Texas Tech Univ.* 146.
- Kartesz, J. T. 1994. A synonymized checklist of the vascular flora of the United States, Canada, and Greenland, 2nd ed. Timber Press, Portland, OR.
- King, W. F., and R. L. Burke (eds). 1989. Crocodilian, tuatara, and turtle species of the world: a taxonomic and geographic reference. Association of Systematics Collections, Washington, DC.
- Krekeler, C. H. 1973. Cave beetles of the genus *Pseudanophthalmus* (Coleoptera, Carabidae) from the Kentucky bluegrass and vicinity. *Fieldiana: Zool.* 62:35–83.
- [KSNPC] Kentucky State Nature Preserves Commission. 1996. Rare and extirpated plants and animals of Kentucky. *Trans. Kentucky Acad. Sci.* 57:69–91.
- [KSNPC] Kentucky State Nature Preserves Commission. 1997. Rare and extirpated plants and animals of Kentucky: 1997 update. *Trans. Kentucky Acad. Sci.* 58:96–99.
- [KSNPC] Kentucky State Nature Preserves Commission. 1999. Rare and extirpated plants and animals of Kentucky: 1999 update. *J. Kentucky Acad. Sci.* 60:124–126.
- [KSNPC] Kentucky State Nature Preserves Commission. 2000. Kentucky natural heritage database, Frankfort, KY.
- McCafferty, W. P. 1996. The Ephemeroptera species of North America and index to their complete nomenclature. *Trans. Am. Entomol. Soc.* 122:1–54.
- Medley, M. E. 1993. An annotated catalog of the known or reported vascular flora of Kentucky. Ph.D. Dissertation. University of Louisville, Louisville, KY.
- Miller, J. Y. (ed). 1992. The common names of North American butterflies. Smithsonian Institution Press, Washington, DC.
- Morse, J. C. 1993. A checklist of the Trichoptera of North America, including Greenland and Mexico. *Trans. Am. Entomol. Soc.* 119:47–93.
- [NMFS] National Marine Fisheries Service. 1999. Endangered and threatened species; revision of candidate species list under the Endangered Species Act. *Federal Register* 64:33466–33468.
- Page, L. M., and B. M. Burr. 1991. A field guide to freshwater fishes. North America north of Mexico. Houghton Mifflin, Boston, MA.
- Paulson, D. R., and S. W. Dunkle. 1999. A checklist of North American Odonata: including English name, etymology, type locality, and distribution. *Slater Mus. Nat. Hist., Univ. Puget Sound Occas. Paper* 56.
- Richter, B. D., D. P. Braun, M. A. Mendelson, and L. L. Master. 1997. Threats to imperiled freshwater fauna. *Conservation Biol.* 11:1081–1093.

- Risk, A. C. 1999. Notes on new and rare mosses to Kentucky. *Evansia* 16:49–58.
- Robins, C. R., R. M. Bailey, C. E. Bond, J. R. Brooker, E. A. Lachner, R. N. Lea, and W. B. Scott. 1991. Common and scientific names of fishes from the United States and Canada, 5th ed. Am. Fish. Soc. Spec. Publ. 20.
- Schuster, G. A. 1997. Description of a new species of terrestrial limnephilid caddisfly (Trichoptera) from Kentucky and West Virginia (USA). Pages 417–424 in R. W. Holzenthal, and O. S. Flint, Jr. (eds). Proceedings of the 8th International Symposium on Trichoptera, 1995. Ohio Biological Survey, Columbus, OH.
- Schweitzer, D. F. 1989. A review of category 2 Insecta in USFWS regions 3, 4, 5. Report prepared for the U.S. Fish and Wildlife Service, Newton Corners, MA.
- Snider, J. A., S. M. Studlar, and M. Medley. 1988. A list of Kentucky bryophytes. *Bryologist* 91:98–105.
- Taylor, C. A., and M. H. Sabaj. 1998. A new crayfish of the genus *Orconectes* from the Blood River drainage of western Kentucky and Tennessee (Decapoda: Cambaridae). *Proc. Biol. Soc. Washington* 111:645–652.
- [TNC] The Nature Conservancy. 1988. Natural heritage program operations manual. The Nature Conservancy, Arlington, VA.
- Turgeon, D. D., J. F. Quinn, Jr., A. E. Bogan, E. V. Coan, F. G. Hochberg, W. G. Lyons, P. M. Mikkelsen, R. J. Neves, C. F. E. Roper, G. Rosenberg, B. Roth, A. Scheltema, F. G. Thompson, M. Vecchione, and J. D. Williams. 1998. Common and scientific names of aquatic invertebrates from the United States and Canada: mollusks, 2nd ed. Am. Fish. Soc. Spec. Publ. 26.
- [USFWS] United States Fish and Wildlife Service. 1992. Endangered Species Act of 1973 as amended through the 100th Congress. Department of the Interior, Washington, DC.
- [USFWS] United States Fish and Wildlife Service. 1994. Endangered and threatened wildlife and plants; animal candidate review for listing as endangered or threatened species, proposed rule. *Federal Register* 59: 58982–59028.
- [USFWS] United States Fish and Wildlife Service. 1999. Endangered and threatened wildlife and plants; review of plant and animal taxa that are candidates or proposed for listing as endangered or threatened; annual notice of findings on recycled petitions; annual description of progress on listing actions; proposed rule. *Federal Register* 64:57533–57547.
- [USFWS] United States Fish and Wildlife Service. 2000. Endangered and threatened wildlife and plants. Department of the Interior, Washington, DC.
- Warren, M. L., Jr. 1992. Variation of the spotted sunfish, *Lepomis punctatus* complex (Centrarchidae): meristics, morphometrics, pigmentation and species limits. *Bull. Alabama Mus. Nat. Hist.* 12.
- Warren, M. L., Jr., W. H. Davis, R. R. Hannan, M. Evans, D. L. Batch, B. D. Anderson, B. Palmer-Ball, Jr., J. R. MacGregor, R. R. Cicerello, R. Athey, B. A. Branson, G. J. Fallo, B. M. Burr, M. E. Medley, and J. M. Baskin. 1986. Endangered, threatened, and rare plants and animals of Kentucky. *Trans. Kentucky Acad. Sci.* 47:83–98.
- Williams, A. B., L. G. Abele, D. L. Felder, H. H. Hobbs, Jr., R. B. Manning, P. A. McLaughlin, and I. P. Farfante. 1989. Common and scientific names of aquatic invertebrates from the United States and Canada: decapod crustaceans. Am. Fish. Soc. Spec. Publ. 17.
- Wilson, D. E., and D. M. Reeder (eds). 1993. Mammal species of the world: a taxonomic and geographic reference, 2nd ed. Smithsonian Institution Press, Washington, DC.

First Observations with the Morehead Radio Telescope, Morehead State University, Morehead, Kentucky

Benjamin K. Malphrus, Michael S. Combs, Michael A. James, D. Shannon Murphy,
and D. Kevin Brown

Morehead Astrophysical Laboratory, Morehead State University, Morehead, Kentucky 40351

Jeff Kruth

Kruth-Microwave Electronics, Ellicott City, Maryland 21043

R. Douglas Kelly

Louisville Male High School, Louisville, Kentucky 40213

ABSTRACT

Herein we report initial astronomical observations made with the Morehead Radio Telescope (MRT). The first radio signals from space were observed with the Morehead Radio Telescope in 1997. The MRT has to date observed a variety of cosmic objects, including galactic sources such as supernova remnants, emission nebula, planetary nebula, extended HI emission from the Milky Way, and the sun, and extragalactic sources such as quasars and radio galaxies. Observations of galactic sources herein reported include Taurus A, Cygnus X, and the Rosette Nebula. Additionally, we report observations of extragalactic phenomena, including Cygnus A, 3C 147, and 3C 146. These initial observations serve as a performance and capability test-bed of the MRT. In addition to the astronomical results of these experiments, tests of the positional accuracy, system sensitivity, and receiver response are inherent in this series of experiments. This paper provides a brief overview of the MRT, including upgrades of major systems, performance characteristics, and a brief discussion of these initial observations.

INTRODUCTION

The Morehead Radio Telescope (MRT), Morehead State University, is an instrument designed by faculty and students of Morehead State University and industrial partners to provide a research instrument for undergraduate astronomy and physics students. The MRT also serves as an active laboratory for physics, engineering, and computer science undergraduates and faculty. The telescope operates in the radio regime at a central frequency of 1420 MHz, which corresponds to the hyperfine transition of atomic hydrogen (HI). The HI spatial distribution and flux density associated with cosmic phenomena can be observed with the instrument. The dynamics and kinematics of objects in space can be investigated by observing over a range of frequencies. The sensitivity and versatility of the telescope design facilitate investigation of a wide variety of astrophysically interesting phenomena. The MRT design provides an instrument capable of supporting scientific research in observational astrophysics at radio frequencies. First light was achieved in October of 1997

with routine observations beginning in January 1998. A brief overview of the current MRT instrumentation, description of major subsystems (antenna, alt-azimuth drive and control systems, receiver systems, and controlling computer and interface), is provided that focuses on instrumentation upgrades at the MRT, followed by a discussion of the first observations and results. A more detailed technical overview of the instrument has been previously published (Malphrus et al. 1998). A brief discussion of the instrumentation is included here, as several major systems have evolved beyond the previously described systems.

The 21 cm Atomic Hydrogen Line

The MRT is designed to operate over a frequency band centered at the 21 cm (1420 MHz) spectral line of atomic hydrogen. Van de Hulst first suggested that the 21 cm line might be detected in the interstellar medium (ISM) in 1945 (Van de Hulst 1945). Unfortunately, Van de Hulst was working in occupied Holland at the time. Searches ensued after

WWII in both the Netherlands and the U.S. The first unambiguous detection of the 21 cm line in the ISM occurred in the U.S. in 1951 and is credited to Ewen and Purcell (Ewen and Purcell 1951). Their experiments were performed with a horn antenna and a sensitive superheterodyne receiver. Subsequently, the HI distribution associated with an ever-increasing number of galactic and extragalactic phenomena have been extensively explored.

The 21 cm emission line arises from a spin-parity reversal of hydrogen in the atomic state that corresponds to a transition between the $F = 0$ and $F = 1$ hyperfine structures of the electron ground state $1^2S_{1/2}$. A transition can occur between the two hyperfine states because they differ slightly in energy, owing to the interaction between the electron spin and the nuclear spin. When the spin parities align, a higher energy state is achieved at $F = 1$. The electron spin parity reversal to the lower hyperfine state occurs naturally on the order of once every 10 million years. Atomic collisions in the radiant medium can greatly accelerate this process to the point of producing a continuous emission. Although the emission frequency can be calculated in terms of well-understood fundamental constants, a correction for the electron spin g factor must be included. Realization that this anomalous factor must be accounted for resulted when comparisons of the 21 cm line first made in 1947 did not agree with the theoretical value. The discrepancy between the theory and empirical values led to the development of quantum electrodynamics (QED). The currently accepted standard frequency of the 21 cm spectral line is 1420.40575186(30) MHz (Storey et al. 1994). The MRT operates a receiver with a 6 MHz bandwidth centered on this frequency.

The science of cm-wave astronomy has provided significant insight into the structure and evolution of cosmic phenomena and afforded a new perspective of the universe. This perspective is considered significant as roughly three-quarters of the material in the universe exists in the form of hydrogen. The MRT scientific goals are predicated upon low resolution, high sensitivity views of the distribution of HI associated with cosmic phenomena.

MRT INSTRUMENTATION

The basic design of the MRT includes a wire-mesh parabolic reflecting antenna, alt-azimuth tracking positioner, control and drive systems, receiver and signal processing system, a controlling computer with an interface device, and supporting electronics and hardware. The system is designed around a total power receiver that converts radiation from space concentrated by the antenna system to an electrical signal, which is amplified, modified and interpreted. The basic measurement that the telescope is capable of is an induced antenna temperature, which is translated into an output voltage at the post-detection stage of the back-end receiver. This voltage corresponds to the total integrated flux density intrinsic to the object over the observed frequency band. A detailed technical overview of the instrument has been previously published (Malphrus et al. 1998). A brief discussion of the instrumentation is included here, as several major systems have evolved beyond the previously described systems.

The MRT system is positioned by a now-second generation system incorporating a controlling computer, optical isolation system, and robotic drive and control systems developed by MSU faculty and students. The current controlling computer is a 450 MHz Pentium II processor with 128 MB of RAM and 9 GB of hard disk space. A multifunction analog, digital, and timing Input/Output (I/O) data acquisition board is installed in the computer. It contains a 12-bit successive approximation A/D converter with 16 analog inputs, two 12-bit D/A converters with voltage outputs, 8 lines of transistor-transistor logic compatible I/O, and two counter/timer channels for timing I/O. The board has a 500kS/s single channel sampling rate. The controlling computer positions the telescope, instructs it in robotic tracking of cosmic sources, and controls data collection and storage. The data from a particular experiment are then transferred via ftp to a Sun Sparcstation or high-end PC for imaging and analysis.

Parabolic Reflecting Antenna

The MRT employs a high-gain 40×11 foot antenna designed for L-Band operation. A

surplused Army NIKE-Hercules ANS-17 Radar antenna was obtained and modified for radio astronomy applications. The antenna was selected because of its relatively large aperture, excellent aperture efficiency (afforded by its innovative offset feed design), and low cost. It includes a parabolic reflector, feed horn and waveguide assembly, and azimuth and elevation positioning system. The positioning system provides azimuth coverage of 360° and elevation coverage of now greater than $0\text{--}90^\circ$. Improvements beyond the previously described system (Malphrus et al. 1998) include the inclusion of a waveguide dehydrator and second-generation digital electronics that drive the positioning system.

MRT Receiver System

The MRT receiver system design and fabrication program was a joint effort between MSU faculty and Kruth-Microwave Electronics Company of Ellicott City, Maryland. The system design is comprised of single receiver with integral low-noise amplifier directly coupled to the waveguide terminus of the MRT antenna system. The overall receiver system design utilizes a low noise, sensitive, stable receiver to convert the 1420 MHz Hydrogen line frequency to a frequency region suitable for processing by standard laboratory equipment. The system is comprised of two major subsystems—the front and back-end receiver systems. The front-end receiver is coupled to the waveguide terminus, which is mounted on the focal feed support of the MRT superstructure. The MRT front-end receiver system incorporates a GaAs FET low-noise amplifier (LNA) design. The back-end IF receiver consists of a processor with four output signals is housed in the Astrophysics Laboratory Control Room. These output signals consist of 3 RF output ports and one DC detector output. The RF output ports provide accessible signals at 160 MHz (6 MHz bandwidth), 21.4 MHz (6 MHz bandwidth), and 21.4 MHz (2 MHz bandwidth). This strategy permits flexibility in signal processing as evolving experimental needs require. The back-end processor also utilizes a frequency synthesizer, associated power supplies, monitor circuitry, and the controlling computer. The DC voltage is derived from an envelope detector and incorporated

into the final stage. Upgrades of the receiver system include a second detector scheme—a digital square law detector, from which the final DC voltage is derived and transmitted to the data acquisition board. All initial observations were made with the MRT receiver system operating in total power mode, using the square law detector.

Performance Characteristics

The MRT primary system performance characteristics have been empirically measured to assist in understanding the results of astronomical observations. The primary system performance characteristics include system temperature, antenna radiation pattern, and antenna gain. The overall system temperature is measured at a respectable 67.3 K (Kruth 1994). The antenna radiation pattern, which is essentially its directivity function on the sky, has been extensively mapped in 2-D and 3-D to determine the main beam and sidelobe structure. These experiments indicated an elliptical beam pattern with a half-power beamwidth (HPBW) of 0.9° and 3.62° for the major and minor axes respectively (Malphrus et al. 1999). This measurement implies good spatial resolution along the horizontal axis of the antenna and less than ideal resolution along the minor axis. The gain of the antenna has been measured at 41dB, which correlates to good sensitivity to objects with relatively low radio frequency flux.

Observations

The performance characteristics of the telescope have made possible observations of a variety of cosmic phenomena. Observations of Taurus A, Virgo A, Cygnus A, Cygnus X, the Rosette Nebula, and two quasars—3C 196 and 3C 147—are herein reported. Analysis of the data is very encouraging in that the transit profiles and maximum voltage deflections measured for objects are comparable to values obtained from observations of the same objects on different days and from observations made with other instruments. Reproducibility in the transit profiles of these objects is apparent. In addition, data taken on consecutive days reveal a sidereal shift in the peaks expected as the earth progresses through the ecliptic plane. The observation techniques are described be-

low. Samples of these data are presented in the Results section.

Three basic modes of observation of celestial objects available with the MRT were used in these experiments: transit, tracking, and mapping. Observing celestial objects in transit mode simply involves pointing the telescope due south (180° azimuth), to the appropriate altitude and observing the object as the rotation of the earth moves (apparently) the object through the telescope's field of view (meridian transit). Observing celestial objects in tracking mode involves using the positioning system of the telescope to compensate for the earth's rotation. The computer controls the telescope's motion as it tracks an object as the object (apparently) moves across the sky. Mapping the HI distribution of a radio source involves scanning the telescope's beam across the source's position repeatedly in a manner similar to the raster scan process utilized to produce television images. Each observational mode is described in some detail below.

Transit Observations

After a target object is selected, the position of the object must be determined as well as the time of meridian transit. Some understanding of coordinate systems is essential to these astronomical observations. The sky operates on the Equator Coordinate System. The MRT operates on the Horizon Coordinate System (as the antenna is alt-azimuth mounted). Therefore, an object's celestial position must be converted to local horizon coordinates to be observed. The values, then, that are important for observing in transit mode are the object's altitude (converted from declination) and local sidereal time of meridian transit.

Determination of transit times in L.S.T. For the given date and zone time (Z.T.) the local sidereal time (L.S.T.) can be calculated for the observer's longitude. L.S.T. is the local hour angle (L.H.A.) of the vernal equinox, that is, the right ascension of the observer's meridian. To calculate L.S.T. from E.S.T. is non-trivial. The following is one of numerous algorithms that may be used:

L.S.T. = K. + 1.0027379(UT) +
(0.06570982 × D) - Longitude/
15°

where:

- UT = Universal time in decimal hours
(UT + 5 hours = E.S.T.)
- D = Number of days since December 31 of the previous year
- Longitude = Geographic west longitude (83° 26^m for Morehead, KY)
- K = 17.369382 (1998);
17.385297 (1999);
17.401211 (2000)

The transit time (time at which an object crosses one's local meridian) may be determined from the object's hour angle. The relationship between sidereal time, hour angle, and right ascension of an object is given:

H.A. = S.T. - R.A.

Transit occurs when the object crosses the local meridian, i.e., when the H.A. = 0; therefore transit occurs when the L.S.T. equals the R.A. of the object. Transit times may be determined by, calculating the E.S.T. of an object when L.S.T. equals the objects R.A. Alternately, an object achieves meridian transit when the L.S.T. is equal to the object's right ascension. Using the Sidereal clock (designed and constructed by students at the Astrophysics Laboratory) to determine transit time simplifies this process.

Determination of transit altitudes. To record the voltage profile of an object at meridian transit, one must know the object's altitude (a) above the local horizon. To find the altitude, one must know the hour angle (H), the declination (δ) of the object, and the geographic latitude of the telescope (φ) must be known. The hour angle of an object at meridian transit by definition is 0. Transit altitudes were calculated using the following equation:

a = sin⁻¹((sin δ)(sin φ)
+ (cos δ)(cos φ)(cos H))

The values of L.S.T. transit and transit altitude are required to position the telescope to observe an object in transit mode. Values obtained for the celestial objects observed are given in Table 1.

Instrumental Procedures

After the coordinate conversion was complete, the MRT Operator program was invoked and the telescope was driven to the ap-

Table 1. Coordinates and flux density values for the initial objects observed with the Morehead Radio Telescope, Morehead State University, Morehead, Kentucky by Malphrus et al. from 1997–2000. Right Ascension and Declination represent fixed values while transit altitude was calculated.

Cosmic object	Right ascension ¹	Declination ²	Transit time (LST)	Transit altitude	RF flux (Jy) ³ (20 cm)
Virgo A (M87)	12 ^h 28 ^m 18 ^s	+12° 40 ^m	12 ^h 28 ^m 18 ^s	64.4°	970
Taurus A	05 ^h 31 ^m 30 ^s	+21° 58 ^m	05 ^h 31 ^m 30 ^s	73.7°	1,420
Rosette Nebula	06 ^h 29 ^m 18 ^s	+04° 57 ^m	06 ^h 29 ^m 18 ^s	56.7°	105
3C 147	05 ^h 38 ^m 43.2 ^s	+49° 49.6 ^m	05 ^h 38 ^m 43.2 ^s	78.4°	58
3C 196	08 ^h 10 ^m 00.1 ^s	+48° 22 ^m	08 ^h 10 ^m 00.1 ^s	79.8°	59
Cygnus A	19 ^h 57 ^m 45 ^s	+40° 36 ^m	19 ^h 57 ^m 45 ^s	87.4°	8,100
Cygnus X	20 ^h 19 ^m 36 ^s	+40° 06 ^m	20 ^h 19 ^m 36 ^s	87.4°	410
Cygnus B	20 ^h 48 ^m 12 ^s	+29° 30 ^m	20 ^h 48 ^m 12 ^s	82.3°	252

^{1,2} Right Ascension and Declination values are taken from the NASA/IPAC Extragalactic Database (NED). NED is operated by the Jet Propulsion Laboratory, California Institute of Technology, under contract with the National Aeronautics and Space Administration.
³ RF fluxes at 20 cm reported are taken from the Third Cambridge Catalog (3C) produced by the Cavendish Laboratory of Cambridge University, UK.

appropriate position for each object. Pre- and post detection gains were set (on the back-end receiver IF processor and square law detector, respectively) based upon published values for the flux density at 20 cm for each of the observed sources (Kraus 1986). These values are reported in Table 1. for each astronomical source. Each source was observed for approximately one hour to provide baseline data before and after each meridian transit. The procedures followed for tracking and mapping runs involve the same initial procedures as those for transit observations, but also include additional, more elaborate procedures described below.

Tracking Observations

Tracking observations involve continuously calculating the object’s azimuth and elevation as time progresses and driving the telescope to the calculated position. The telescope can also be “manually” driven from the back-end with the drive and control system to maintain “peak on source”. A combination of using the computer-generated coordinates and nudging the telescope with the manual drive system has proven most effective thus far. All peak voltages herein reported were measured during tracking observations utilizing a variety of integration times.

Mapping Observations

Observing with the MRT in mapping mode is the most challenging but rewarding observational procedure. A pre-determined area

(usually rectangular) is scanned with the telescope beam as the object is tracked across the sky. The target area is scanned in a manner similar to the raster scan process utilized by monitors and television sets to produce images. A single elevation is scanned over a pre-determined range of azimuths in one direction (i.e., CW). The telescope is then driven to a lower elevation (typically 1°) and the same range of azimuth values is scanned by driving the telescope in the opposite (CCW) direction. This process is repeated until the entire target area is scanned. 2-D (contour) and 3-D (topographic) maps of the HI associated with the astronomical object can be produced with these data. These maps represent the spatial distribution of HI in the target region (integrated over the entire receiver post-detection bandwidth). The data reduction and imaging software utilized in these experiments is the HiQ package developed by National Instruments. Additional software that allows the observer to reduce and edit the data as well as transform the raw data into an appropriate matrix format was developed by MSU students. A more detailed description of the mapping procedure, including the details of data reduction, is described in a later publication.

RESULTS

To date, observations of a variety of cosmic phenomena have been undertaken. Herein, we describe observations of Virgo A, the Cygnus A Complex, Taurus A, the Rosette Nebula, 3C 196, and 3C 147. Telescope receiver

Table 2. Telescope settings and results obtained for the initial astronomical observations made with the Morehead Radio Telescope, Morehead State University, Morehead, Kentucky by Malphrus et al. from 1997–2000.

Cosmic object	Pre-detection gain	Post-detection gain	Integration time (s)	TOS	ΔV_{max}	N_{rms}	S/N
Virgo A (M87)	40%	900x	1 ^s	2160s	4.1v	0.3v	13.6/1
Taurus A	40%	900x	10 ^s	864s	4.9v	0.1v	49/1
Rosette Nebula	40%	1000x	1 ^s	684s	5.7v	0.05v	114/1
3C 147	40%	1000x	1 ^s	1200s	3.8v	1.5v	2.5/1
3C 196	40%	1000x	1 ^s	1200s	5.8v	0.3v	19.3/1
Cygnus A	50%	900x	1 ^s	2400s	2.5v	0.1v	25/1
Cygnus X	50%	900x	1 ^s	1080s	0.5v	0.1v	5/1
Cygnus B	50%	900x	1 ^s	2400s	0.3v	0.1v	3/1

settings, integration times, and other observational parameters such as time on source are listed in Table 2. Also listed are induced voltages, and calculated signal-to-noise ratios. Pre-detection gain, post-detection gain, and integration time are telescope values set by the observer. Time on source (TOS) is determined by the spatial extent of the object when observed in transit mode. V_{max} , N_{min} , ΔV , and S/N represent experimental results. V_{max} represents the maximum or peak voltage induced by the cosmic object and is proportional to the flux density of the object as explained below. N_{rms} is the baseline width, which also corresponds to the rms noise temperature of the system. ΔV is the peak voltage minus the averaged baseline voltage level.

The peak voltage ΔV , the basic measurement in total power mode is a function of the induced antenna temperature (ΔT). The antenna and front- and back-end receivers of a radio telescope system together act as a radiometer for measuring the temperature of distant regions of space coupled to the system through the radiation resistance of the antenna. The temperature of the radiation resistance is determined by the temperature of the emitting region seen by the beam of the antenna as defined by its directivity function, i.e., its radiation pattern. The temperature of the antenna radiation resistance is referred to as induced antenna temperature. A mathematical expression describing induced antenna temperature demonstrates that the induced temperature is a function of the cosmic radiator convolved with the antenna's directivity function (Kraus 1986):

$$\Delta V \approx k\Delta T = w$$
$$= \frac{1}{2}A_e \iint B(\theta, \phi)P_n(\theta, \phi) d\Omega$$

where:

ΔV = induced post-detection voltage, volts

ΔT = induced antenna temperature, K

w = received power per unit bandwidth, $w \times \text{Hz}^{-1}$

A_e = effective aperture (physical aperture \times aperture efficiency), m^2

$B(\theta, \phi)$ = source brightness distribution, dimensionless

$P_n(\theta, \phi)$ = antenna radiation pattern, dimensionless

$d\Omega = \sin\theta d\theta d\phi$ = element of solid angle, rad^2

k = Boltzman's constant $1.38 \times 10^{-23} \text{ J} \times \text{K}^{-1}$

Given that the cosmic object's energy flux density is also a function of the cosmic radiator's brightness convolved with the antenna's directivity function (Kraus 1986)

$$\Delta S = \iint_{\text{Source}} B(\theta, \phi)P_n(\theta, \phi) d\Omega,$$

where:

ΔS = total source flux density, $w \times \text{m}^{-2} \times \text{Hz}^{-1}$

$B(\theta, \phi)$ = source brightness distribution, dimensionless

$P_n(\theta, \phi)$ = antenna radiation pattern, dimensionless

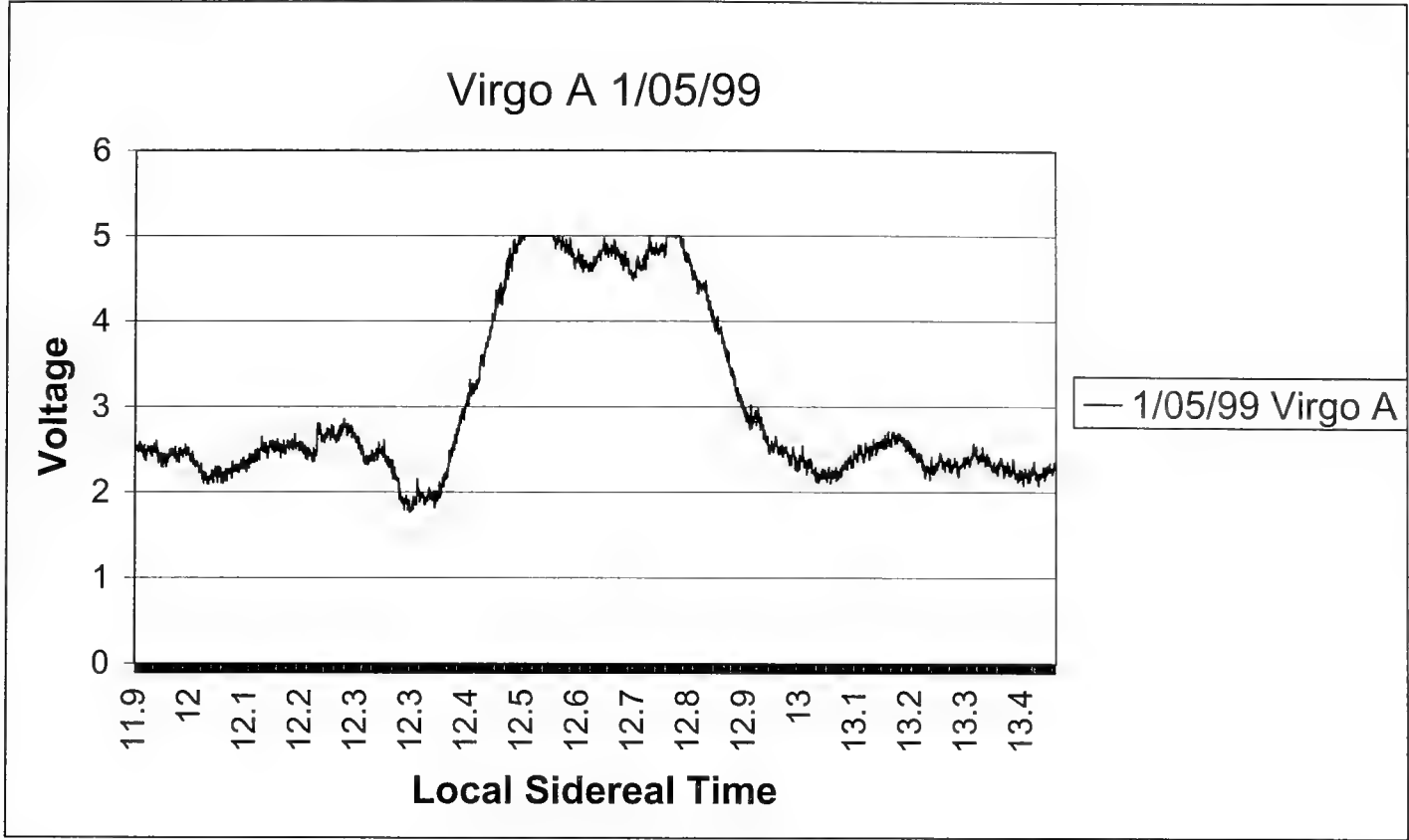


Figure 1. Morehead Radio Telescope, Morehead State University, Morehead, KY. A transit observation of Virgo A taken with the MRT on January 5, 1999.

$$d\Omega = \sin\theta \, d\theta \, d\phi = \text{element of solid angle, rad}^2$$

It follows that ΔS simplifies to

$$\Delta S = \frac{2k\Delta T}{Ae}.$$

A relative value as opposed to an absolute value for ΔT , and therefore ΔS was obtainable during these experiments. ΔT can be calibrated against a standard induced voltage or astronomical flux calibrator in future experiments. The S/N_{rms} , however, can be measured from the voltage profiles because ΔV_{max} corresponds to the signal (S), and N_{rms} , the rms noise corresponds to the baseline width. The calculated S/N values for each observation are given in Table 2. The S/N is expressed as a ratio. A variety of graphical representations of the data as well as a brief description of the astronomical objects observed are provided below.

Virgo A

Virgo A is the fifth brightest radio object in the sky, with a flux density of 970 Jy. The ob-

ject is a radio galaxy with some strange features that make it highly visible in the radio spectrum. Virgo A has an odd jet that extends from the west-side of the nucleus. This jet, apparent at extremely high spatial resolutions, is thought to be associated with material ejected from the nucleus. The system is an extremely powerful emitter of both waves and X-rays. A transit observation of Virgo A taken with the MRT on 5 Jan 1999 is provided in Figure 1.

Taurus A

Taurus A is a supernova remnant (SNR) in the constellation Taurus; it is also known as the Crab Nebula. Taurus A is located about 6300 light years away and is the fourth brightest radio object in the sky. It has a flux density of 1420 Jy. Successive transit observations of Taurus A were made on 19–20 Aug 1997. These observations are provided in Figure 2. An expected sidereal shift of 4 minutes of time in the transit peaks is observed.

The Rosette Nebula

The Rosette nebula is a supernova remnant (SNR) within our galaxy that has some inter-

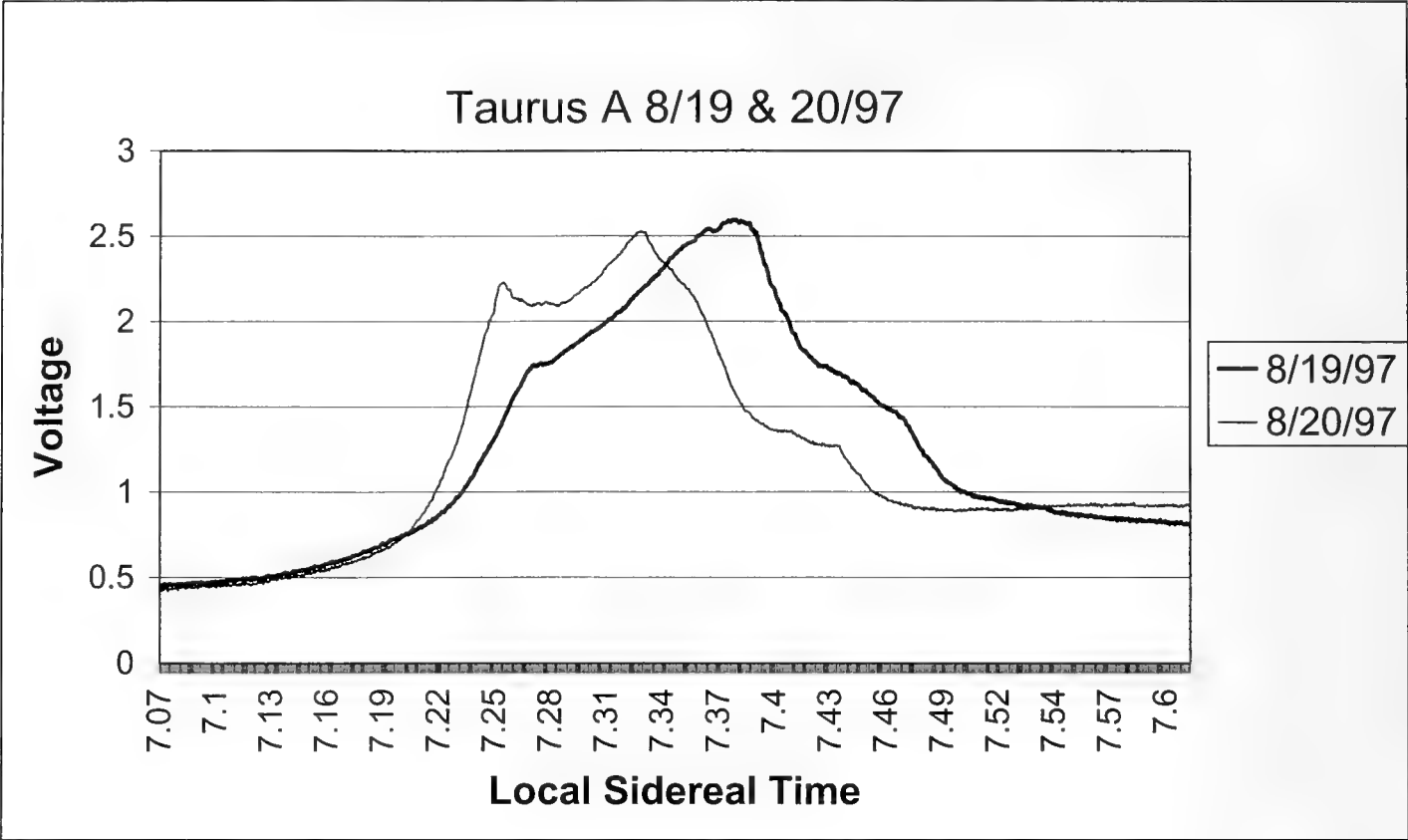


Figure 2. Morehead Radio Telescope, Morehead State University, Morehead, KY. Successive transit observations of Taurus A made on August 19th and 20th, 1997.

esting features. It is also classified as an emission nebula—a cloud of high temperature gas. The atoms in the cloud are energized by ultraviolet light from a nearby star and emit radiation as they fall back into lower energy states; hence we can see the Rosette Nebula in the radio regime. Emission nebulae are usually sites of star formation; the Rosette Nebula does in fact have star formation occurring in its outer regions. A transit observation made of the Rosette Nebula is shown in Figure 3.

3C 147

3C 147 (also known as [HB89] 0538 + 498 and QSO OG +465) is a distant quasar that has been previously observed by numerous investigators. The quasar is extremely distant and faint in the optical spectrum exhibiting a visual magnitude of 17.8 and a redshift of $z = 0.54500\ 79$ (NASA/IPAC Extragalactic Database (NED), 1999). It is optically variable and unresolved by most instruments. The object shows an unusually complex, nonlinear structure that varies with time. Superluminal sep-

aration of two components in the core region has been observed. A jet is embedded in the diffuse emission region. VLA images at 1 GHz indicate a weak component north of the main component opposite the jet with respect to the core. Although the object is spatially unresolved by the MRT, its luminous output in the radio spectrum is well above the detection limits of the instrument. Figure 4 depicts a transit observation of 3C 147 in which a favorable signal to noise ratio of 2.5:1 was achieved.

3C 196

3C 196 (also known as [HB89] 0809+483) is a distant quasar in the constellation Lynx. It is an extremely distant and faint object, exhibiting a redshift of $z = 0.87100$ and an apparent visual magnitude of 17.79 79 (NASA/IPAC Extragalactic Database (NED) 1999). The object has a famous and well-studied absorption system at $z = 0.43685$, which gives rise to a host of metal line species. The 21 cm absorption is especially significant because it occurs in a resolved background source, which allows useful

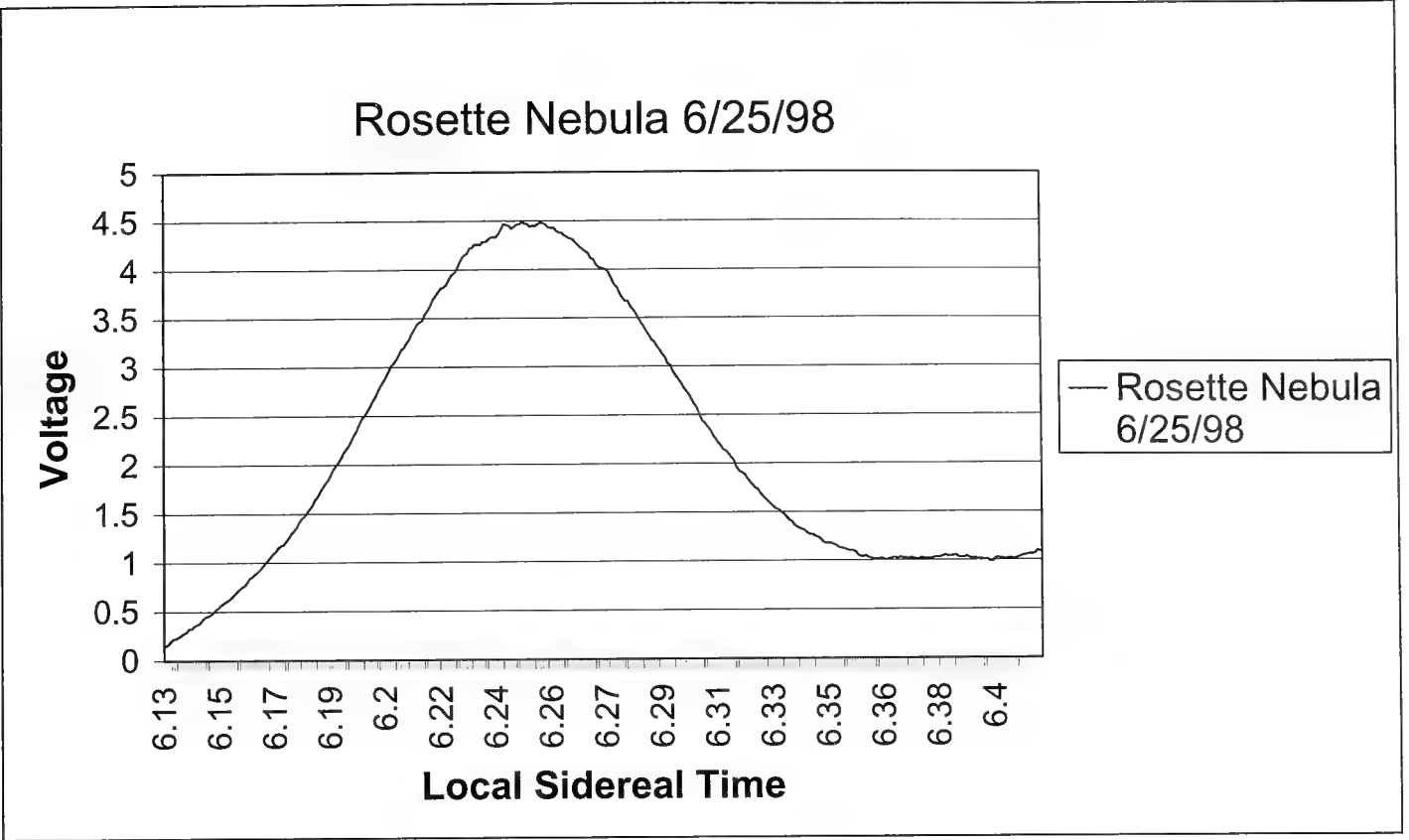


Figure 3. Morehead Radio Telescope, Morehead State University, Morehead, KY. A transit observation of the Rosette Nebula taken with the MRT on June 25, 1998.

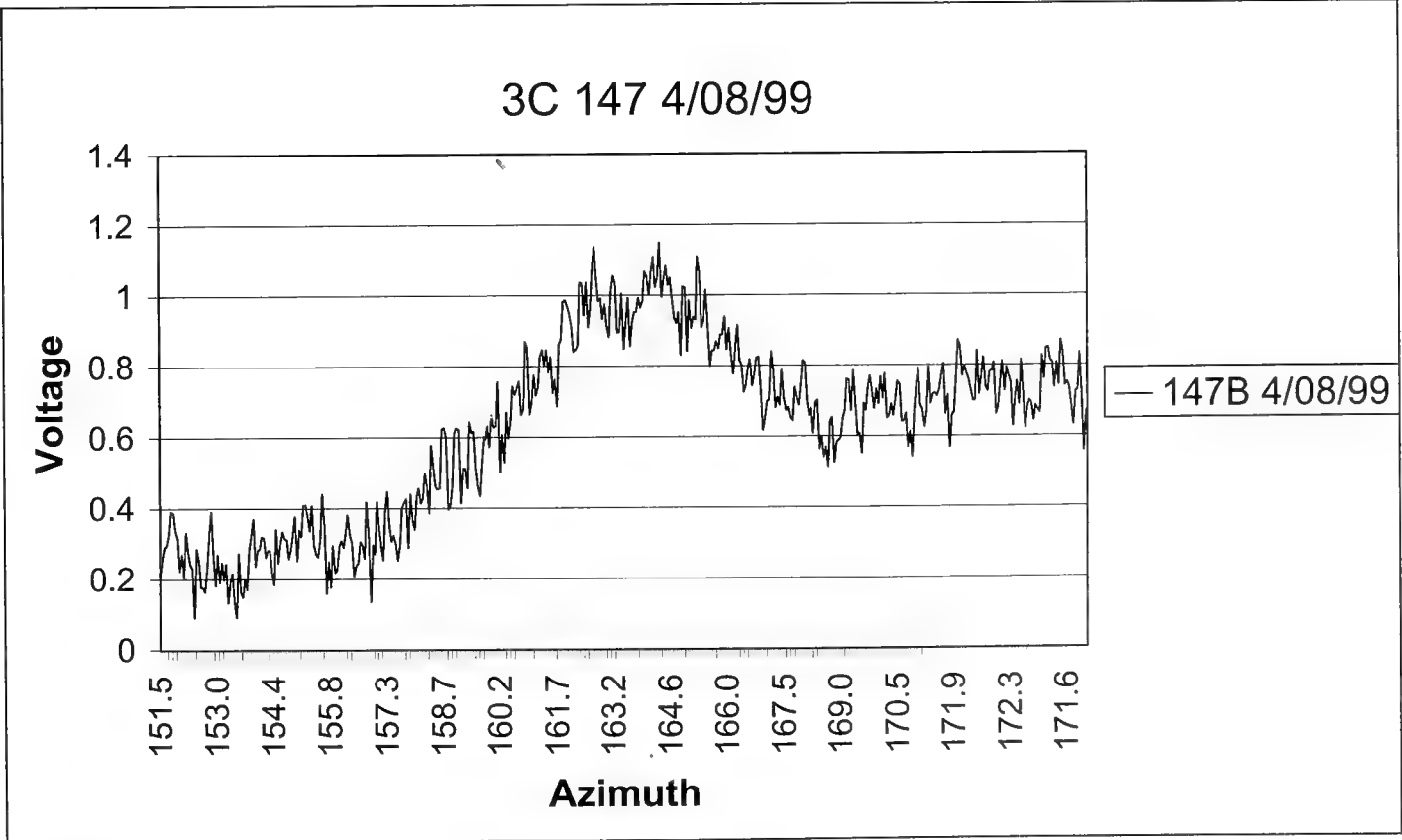


Figure 4. Morehead Radio Telescope, Morehead State University, Morehead, KY. A scan observation made of the quasar 3C 147 on April 8, 1999.

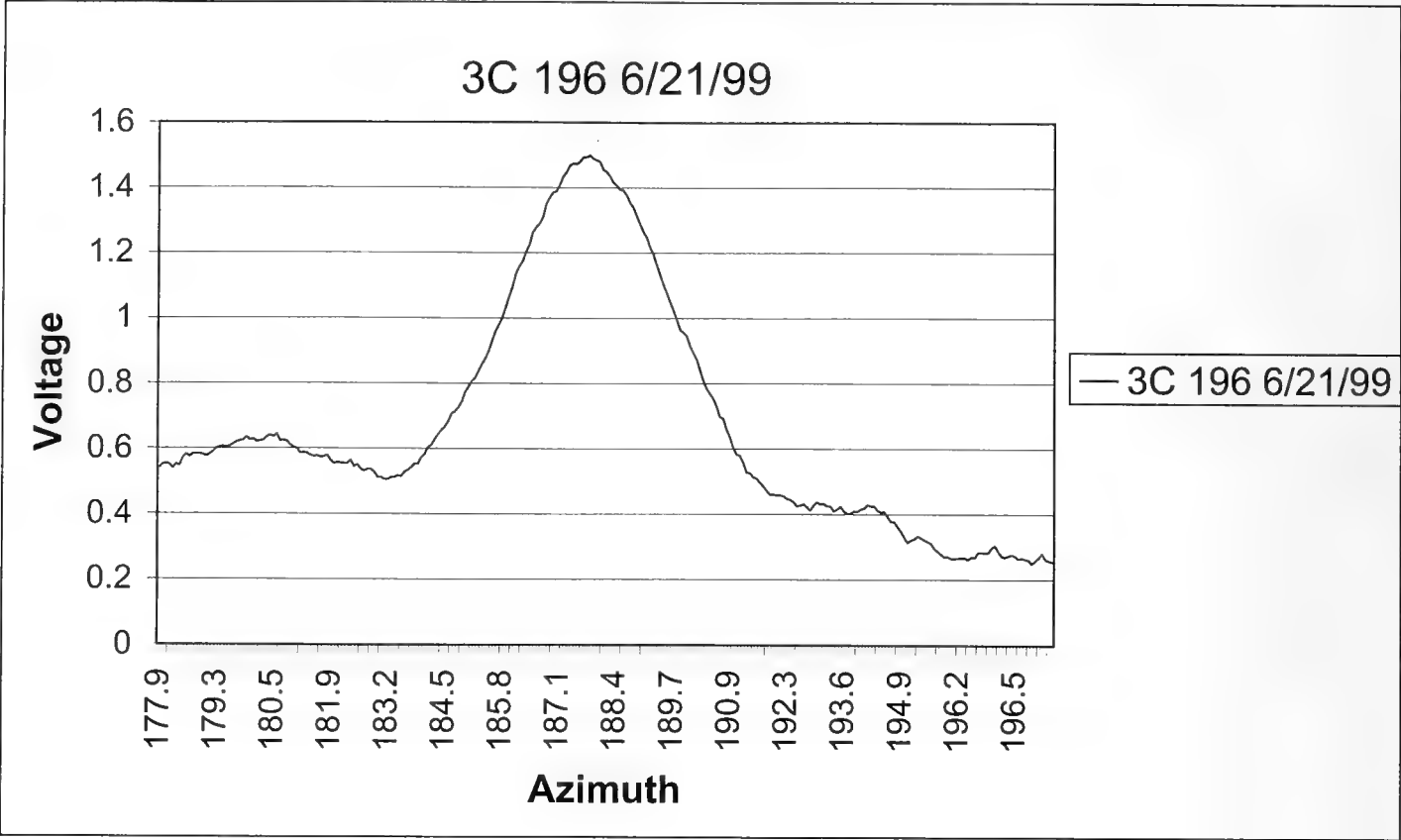


Figure 5. Morehead Radio Telescope, Morehead State University, Morehead, KY. A scan observation made of 3C 196 on June 21, 1999.

limits to be placed on the absorber size. There is also an associated absorber in this object at $z = 0.8714$ with an apparent infall velocity of 100 kmsec^{-1} . A transit observation made with the MRT of 3C 196 is presented in Figure 5.

The Cygnus A Complex

The Cygnus A Complex, a very active region in the radio sky, contains at least three major components, Cygnus A, Cygnus B, and Cygnus X. Cygnus A is a distant radio galaxy approximately 1 billion light years away. It is one of the best-known radio sources in the sky but it has no bright visible object that corresponds to the radio emission. No visible object was associated with the radiation until 1951 when astronomers at the Palomar Observatory found an object that appeared as a pair of unresolved 18th magnitude objects. Cygnus A is the second most luminous radio object that is observable, with a flux density of 8100 Jy , second only to Cassiopeia A. Cygnus A represents the largest peak in the topographical representation of the data. Cygnus X, the second strongest peak in the data, is associated with

the famous Cygnus X black hole. The radio waves emanate from the accretion disk associated with the black hole. The accretion disk is produced as the black hole's gravity well accretes matter from its companion star, swirling it into a flattened disk and heating the infalling material to 10^6 K , causing it to radiate in the X-ray and radio regions. Cygnus X is considered among the most convincing candidates for a galactic black hole. Since Cygnus X lies close to the line of sight of Cygnus A, it is almost impossible to observe one object and not the other in a transit observation. Since these objects lie close to each other in line of sight, but not in physical space, their combined observable radiation is called the Cygnus A Complex. We interpret the component labeled Cygnus B in the data to be associated with the Cygnus Loop, an ancient supernova remnant in the galaxy some 770 pc distant from earth. A transit profile of the Cygnus complex as well as three other perspectives of the HI distribution as produced with the MRT is shown in Figure 6. Complex structural de-

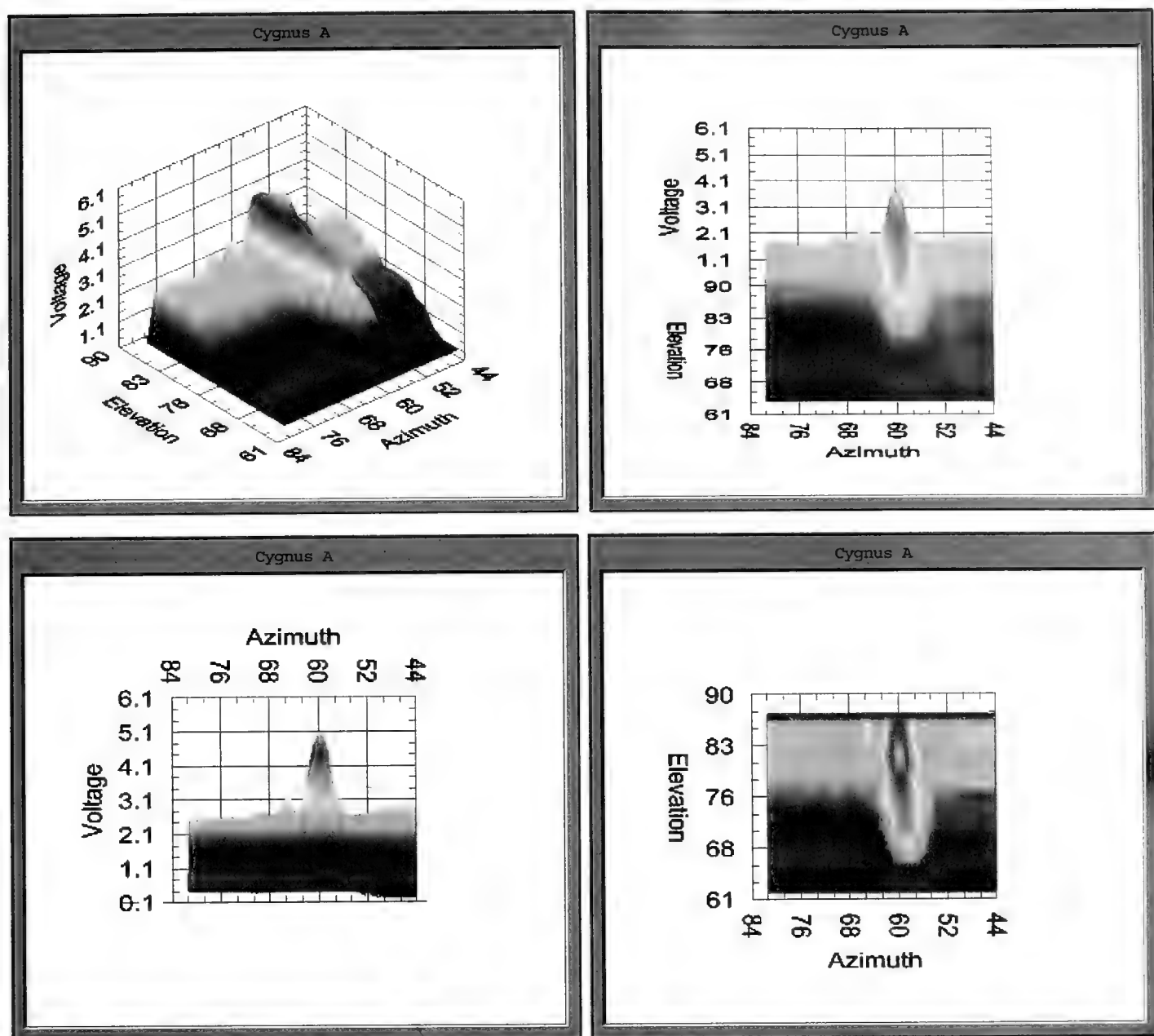


Figure 6. Morehead Radio Telescope, Morehead State University, Morehead, KY. Four perspectives of a map of the spatial distribution of HI associated with the Cygnus A, B, and X radio sources.

tail is evident in the HI associated with these regions.

DISCUSSION

Results of these initial observations are very promising. The HI profiles and distributions compare favorably to transit profiles and HI distribution maps produced with comparable instruments. Transit profiles of the Cygnus A Complex and Virgo A are extremely similar in structure to ones produced with the NRAO Green Bank 40-foot Radio Telescope and the University of Indianapolis 5-meter antenna. The HI distribution map compares very fa-

vorably to those produced with the NRAO Green Bank 40-ft. Radio Telescope and the Ohio State University "Big Ear." These HI profiles and maps compare favorably to previously produced images despite the MRT's rather elliptical beam.

Results of these initial observations are encouraging in that they attest to the performance characteristics of the MRT. The reproducibility apparent in the data attests to the MRT receiver stability and pointing accuracy. The results of these initial experiments are very exciting in that the capabilities of the instrument to perform more extensive and per-

spicacious experiments is realized. Challenges still exist, however, in the procedural elements of each observing mode. The results of observations in transit mode are haunted by the elliptical, vertically-oriented beam. Circular beams are ideal for these type experiments. Observations made in scanning mode are challenging in that it is difficult to scan the telescope at a constant rate and produce the same number of data points for each scan. Reducing the data and preparing the data matrix for 3-D maps is complicated by this problem. In tracking mode, there are the inherent problems of pointing accuracy. Even still, these early observations are encouraging and indicate that the MRT systems have, even at this formative stage, exceeded expectations.

A limitation of the current data is that the observations are essentially uncalibrated. Future experiments may attempt to "bracket" observations of the astronomical sources with observations of known flux calibrators, a strategy utilized at the National Radio Astronomy Observatory's Very large Array (VLA). Other possibilities include injecting a noise source of a standard voltage directly into the waveguide to coaxial transition via a hardware (calibration) switch. This strategy is employed with the National Radio Astronomy Observatory's 40-foot Radio Telescope. Still another possibility involves generating a test (calibration) signal with an RF generator in the Laboratory Control Room and coupling it to a quasi-directional test antenna. Observations of the astronomical sources could then be bracketed by observations of the calibration signal.

Next Experiments

A next generation of experiments is indicated as a result of analysis of these early data. MRT systems experiments to be performed include determining the electrical focus of the antenna system, determining the mechanical focus of the antenna system, empirically determining the minimum detectable flux density, and mathematically modelling the antenna surface. The future, in terms of the possibilities of astronomical observations, is very exciting. The instrument is capable of observing many of the most exotic and energetic astronomical phenomena in the universe. Astronomical investigations include observations of

a wider variety of cosmic sources (including such galactic objects as black holes, pulsars, supernova remnants, starbirth nebulae, and extragalactic objects such as active galaxies, radio galaxies, and interacting galaxy systems), the production of a map of the HI distribution associated with the Milky Way, an all-sky northern hemisphere map, and SETI projects.

ACKNOWLEDGMENTS

The Morehead Radio Telescope Project was funded by the National Science Foundation under the Instruments and Laboratory Improvements Program, NASA and by Morehead State University. The MRT receives continued support from Morehead State University's College of Science and Technology, Department of Physical Sciences and Department of Industrial Education and Technology and from Kruth-Microwave Electronics of Hanover, Maryland, and Dan Puckett Engineering, Morehead, Kentucky. This research has made use of the NASA/IPAC Extragalactic Database (NED), which is operated by the Jet Propulsion Laboratory, California Institute of Technology, under contract with the National Aeronautics and Space Administration.

LITERATURE CITED

- [ARRL] The ARRL antenna book. 1998, 18th ed. The American Radio Relay League, Newington, CT.
- Ewen, H. I., and E. M. Purcell. 1951. Observation of a line in the galactic radio spectrum. *Nature* 168(4270): 356-358.
- Kraus, J. D. 1986. *Radio Astronomy*, 2nd ed. Cygnus-Quasar Books, Powell, OH.
- Kruth, J. 1994. A receiver system for radio astronomy. Kruth Microwave Corporation. Internal Document, Ellicott City, MD.
- Malphrus, B. K., M. S. Combs, R. P. Lillard, and J. Kruth. 1999. Empirical measurements of the antenna radiation pattern of the Morehead Radio Telescope. *J. Kentucky Acad. Sci.* 60:113-123.
- Malphrus, B. K., E. Thomas, M. S. Combs, B. Ratliff, B. Roberts, C. Pulliam, J. Carter, D. Preece, V. Hullur, R. Brengelman, D. Cutts, C. J. Whidden, R. Stanley, W. Grizes, D. Henderson, D. Puckett, and J. Kruth. 1998. The Morehead Radio Telescope: design and fabrication of a research instrument for undergraduate faculty and student research in radio frequency astrophysics. *J. Kentucky Acad. Sci.* 59:77-92.

- Malphrus, B. K. 1996. The history of radio astronomy and the National Radio Astronomy Observatory: evolution toward big science. Krieger Publishers, Melbourne, FL.
- Storey, J. W., M. C. B. Ashley, M. Naray, and J. P. Lloyd. 1994. 21 cm line of atomic hydrogen. *Am. J. Phys.* 62(12):1077–1081.
- [NED] NASA/IPAC extragalactic database. Jet Propulsion Laboratory, California Institute of Technology, under contract with the National Aeronautics and Space Administration, Pasadena, CA.
- Van de Hulst, H. C. 1945. Radio waves from space. *Ned. Tijdschr. Natuurk.* 11:201–202.

***Agrimonia* (Rosaceae) in Kentucky with Notes on the Genus**

Mark V. Wessel and John W. Thieret

Department of Biological Sciences, Northern Kentucky University, Highland Heights, Kentucky 41099

ABSTRACT

The genus *Agrimonia* (Rosaceae) consists of about 15 species worldwide, 7 in the U.S., and 4 in Kentucky: *A. gryposepala*, *A. parviflora*, *A. pubescens*, and *A. rostellata*. The reported presence in Kentucky of three additional species—*A. eupatoria*, *A. microcarpa*, and *A. striata*—could not be verified. Included are a key to and descriptions of the Kentucky taxa and notes on biology and uses of *Agrimonia*.

INTRODUCTION

The genus *Agrimonia* (Rosaceae), with about 15 species, occurs in North America, South America, Eurasia, and South Africa. Because of the problems of identification of some species of the genus and the uncertainty concerning which species actually occur in Kentucky, we decided to examine the Kentucky species to determine their taxonomy and distribution. Additionally we began a survey of previous work on *Agrimonia*, presenting here stray notes—not an exhaustive literature review—on the genus.

AGRIMONIA IN KENTUCKY

Species of *Agrimonia*—the agrimonies—were early recorded as components of the Kentucky flora. M'Murtrie (1819), in his *Flora Louisvilleensis*, listed three: *A. eupatoria* (a European taxon, almost certainly a misidentification for one of the native Kentucky species), *A. parviflora*, and *A. sylvatica* (the last mentioned name we have as yet been unable to trace). Short, Peter, and Griswold's (1833) catalog of the phaenogamous [sic] plants of Kentucky—which, so far as we are aware, is the earliest attempt to account for the flora of a U.S. state—has two: *A. eupatoria* and *A. parviflora*. The following year Short and Peter (1834) changed *A. parviflora* to *A. suaveolens* (the latter now regarded as a synonym of the former). According to Medley (1993), Short's specimens labelled *A. eupatoria* and deposited at PH have been annotated as *A. striata*, an identification we regard as suspect. Kentucky specimens collected in the 1830s by either Short or Peter and sent to us on loan from PH are *A. parviflora*, *A. pubescens*, and *A. rostellata*; *A. striata* was not represented. Price (1893) had only *A. eupatoria* for Warren

County; almost certainly this too is a misidentification. Half a century later McFarland (1942) listed the four species that we recognize as representing the genus in Kentucky; Braun (1943) listed only three, not including *A. gryposepala*. Browne and Athey (1992) listed seven species for the state: the four we recognize in our paper and three others—*A. eupatoria*, *A. microcarpa*, and *A. striata*—the occurrence of which in Kentucky we have not been able to confirm; the listing of these three is, we believe, based on misidentification. In the most recent summary of the Kentucky flora Medley (1993) accepted for the state *A. gryposepala*, *A. microcarpa*, *A. parviflora*, *A. pubescens*, and *A. rostellata*.

MATERIAL AND METHODS

The data herein are based on 421 herbarium specimens (ca. 12% misidentified) of Kentucky *Agrimonia* borrowed from 14 herbaria; on the 46 collections of *Agrimonia* we and colleagues made from 1996 through 1999 in 18 counties of Kentucky (specimens in KNK); and on previously published accounts of the genus (Bicknell 1896, 1901a, 1901b; Bush 1916; Fernald 1950; Gleason and Cronquist 1991; Kline and Sørensen 2000; K.R. Robertson 1974; Robinson 1900, 1901; Rydberg 1913; Skalický 1973; Svenson 1941; Torrey and Gray 1838–1840). From herbarium sheets we recorded the following data: county, location, collector(s), date; leaf length; leaflet count, length, and width; sepal length; hair length; and fruit length and width ("fruit" = fruiting hypanthium with enclosed achene(s) but excluding bristles and sepals).

When collecting specimens of *Agrimonia*, one should be certain to obtain the roots, which are helpful in identification, and, if pos-

sible, mature, fully reflexed fruits that are at least half mature.

We saw no specimens of *Agrimonia* from 35 of Kentucky's too many (120) counties: Adair, Boyd, Carroll, Christian, Clay, Cumberland, Elliott, Fulton, Graves, Grayson, Hancock, Harrison, Henderson, Hopkins, Johnson, Knott, Knox, Leslie, Livingston, Logan, Martin, Mason, Mercer, Metcalfe, Monroe, Owsley, Perry, Russell, Scott, Shelby, Simpson, Trimble, Union, Washington, and Webster.

TAXONOMY

In North America *Agrimonia* can easily be distinguished from other, sympatric genera of the Rosaceae by its herbaceous habit, its five yellow petals, its five sepals, and its hypanthium ("fruit") armed with hooked bristles. To identify *Agrimonia* correctly, one must learn to distinguish the different types of stem vestiture (see Jain and Singh [1973] for a study of the hairs of *A. eupatoria*): (1) minute gland-tipped hairs, the stalk short, few-celled; (2) long, straight or curved, eglandular hairs 1.0–4.0 mm; and (3) short, often curved, sometimes matted, eglandular hairs < 0.5 mm.

Our species of *Agrimonia* flower from July to August or September. Individuals are rare to frequent, but never, in our experience, common or abundant.

Figures 1 and 2 are of *Agrimonia parviflora*, a representative member of the genus.

Agrimonia Linnaeus

Herbs perennial, erect, hemicryptophytic, with crystals of calcium oxalate in parenchymatous tissues (Murata and Umemoto 1983). **Roots** fibrous, sometimes with fusiform tubers. **Stems** branched or unbranched. **Leaves** alternate, stipulate, odd 1-pinnate, of 2 main sizes, major (larger) leaflets 3–19(21), toothed, terminal leaflet often the largest, minor (smaller) leaflets 0–37, sometimes bractletlike, interspersed among major leaflets. **Inflorescence** racemose, terminal and often axillary, few to many flowered; flowers nearly sessile, pedicels bracteate at base, ascending, stipe spreading or reflexed in fruit. **Flowers** subopposite to alternate, 4–9 mm in diameter, bi-bracteolate, perigynous; sepals 5, persistent, forming a beak in fruit; petals 5, yellow (very rarely white); stamens 5–20; pollen 3-colporate; ovaries 2, enclosed by hypanthium, styles

exserted. **Fruit** (i.e., fruiting hypanthium plus enclosed achene[s] but excluding bristles and sepals) constricted at the throat, indurate, hemispheric to top-shaped, stipitate, rim bearing erect to reflexed, hooked bristles; achenes 1 or 2. Type species: *Agrimonia eupatoria* L. For illustrations of this species see Phelouzat (1963) and Ross-Craig (1956).

Key to Kentucky Species of *Agrimonia*

The l:w data in the key and in each description below refer to the average length:width ratio (plus range of values) of the blade of the largest terminal leaflet on a plant.

1. Major leaflets mostly 9–19; l:w = 4.0 (3.3–4.8) *A. parviflora*
1. Major leaflets mostly leaves 3–9; l:w < 3.0 2
2. Rachis of inflorescence copiously pubescent, eglandular or nearly so or the glands concealed by hairs; leaflets downy pubescent beneath; roots with fusiform tubers *A. pubescens*
2. Rachis of inflorescence glandular, pubescence sparse or absent; leaflets glandular beneath, glabrous to sparsely hirsute along veins; roots with or without fusiform tubers 3
3. Rachis of inflorescence with glands and long spreading hairs to 2 mm; fruits more than 6 mm, the bristles to 4.0 mm; sepals in fruit 2–3 mm; roots without fusiform tubers *A. gryposepala*
3. Rachis of inflorescence with glands only or also with a few hairs to 1 mm; fruits less than 5 mm, the bristles 2 mm or less; sepals in fruit 1.5–1.8 mm; roots with fusiform tubers *A. rostellata*
1. ***Agrimonia gryposepala*** Wallr. [Greek *grypos*, curved, and New Latin *sepalum*, sepal] Figures 3, 7, 11

Herb 3–18 dm, roots without tubers. **Stem:** vestiture of 2 types: (1) minute, gland-tipped hairs; and (2) long, stiff hairs to 3.2 mm. **Leaves:** to 21 cm; major leaflets 3–9, ovate or obovate to elliptic or rhombic, 1–10.5 × 0.8–5.2 cm, adaxially glabrous or nearly so or with a few scattered long hairs, abaxially with



Figure 1. *Agrimonia parviflora*, a representative species of the genus, $\times \frac{1}{2}$. From Zardini 1971, with permission.

gland-tipped hairs and with long hairs 0.5–2 mm; l:w = 2.0 (1.8–2.5); minor leaflets 0–9, to 2.3 cm, 0–3 pairs between major leaflets. **Inflorescence** abundantly glandular and with sparse, long, stiff hairs to 2 mm. **Fruit** (excluding bristles sepals) 6.8–7.5 \times 3.5–5.8 mm, glandular, the hypanthium top-shaped to campanulate, deeply grooved, the ridges, base, and pedicel with a few scattered, stiff, white hairs to 0.5 mm, the grooves glandular, otherwise glabrous; sepals in fruit 2–3 mm; bristles 2.5–3.7(4.0) mm, in 4–5 rows, lowermost row sharply reflexed. 2n = 56 (Brittan 1953).

Woodland margins, thickets, clearings, fields, and disturbed sites. *Agrimonia gryposepala* is rare and of limited occurrence in Kentucky. Harlan is the only Kentucky county from which we have seen a herbarium specimen to document the occurrence of the species in the state. *Agrimonia gryposepala* is considered a threatened species in Kentucky (KSNPC 1996). Kline and Sørensen (1990) discussed lectotypification and synonymy of this species.

2. ***Agrimonia parviflora*** Sol. In Ait. [Latin

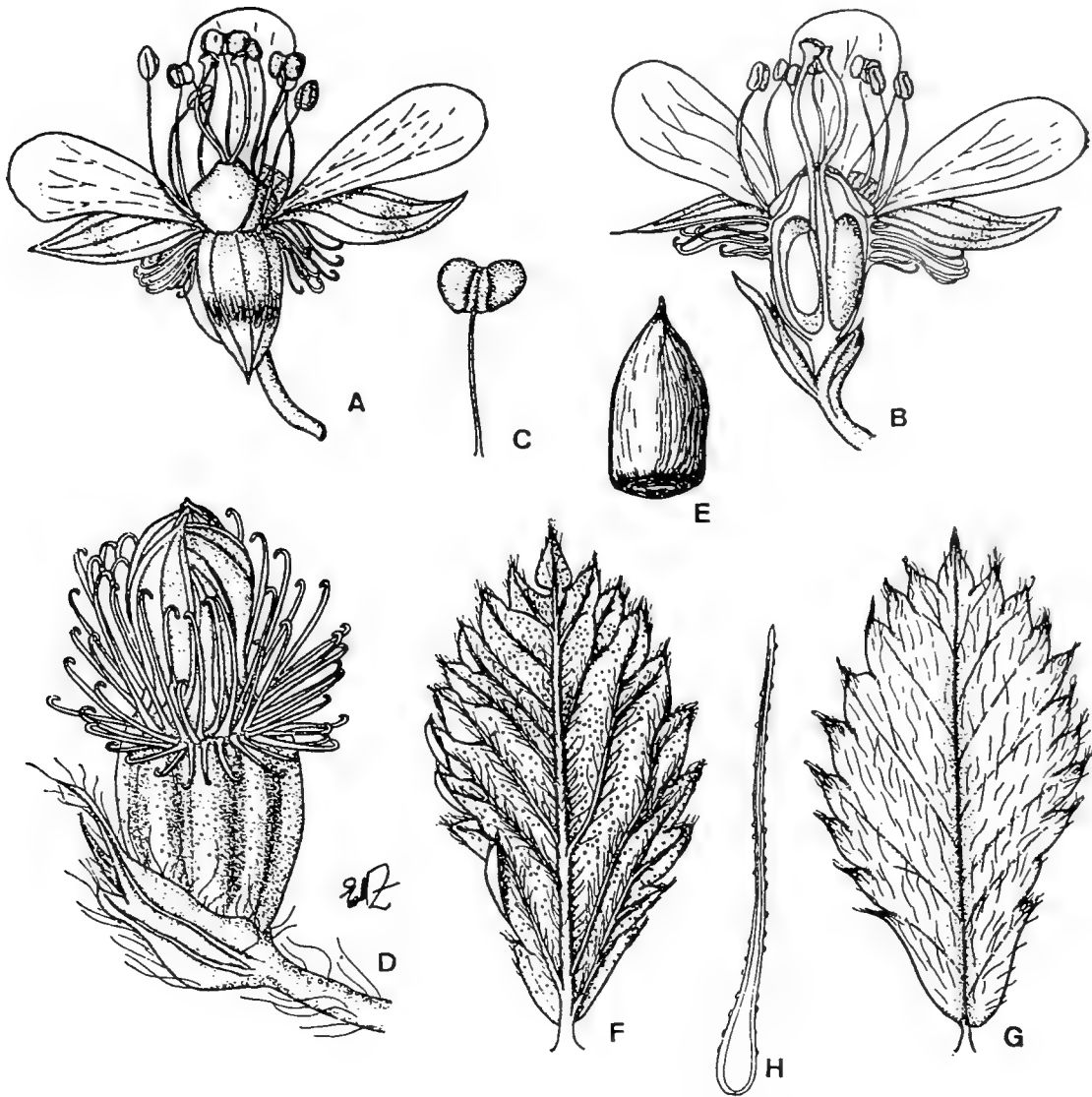


Figure 2. *Agrimonia parviflora*. Details of flower and leaflet. (A) Flower with 2 forward petals removed, $\times 10$. (B) Longitudinal section through flower, with bracteoles, $\times 10$. (C) Stamen, $\times 15$. (D) "Fruit," with bracteoles, $\times 10$. (E) Achene, $\times 10$. (F) Leaflet, adaxial surface, $\times 2$. (G) Leaflet, abaxial surface, $\times 2$. (H) Trichome from abaxial surface of leaflet, $\times 20$. From Zardini 1971, with permission.

parvus, small, and *flos*, flower, alluding to size of the flowers] Figures 1, 2, 4, 8, 12, 15

Herb 5–20 dm; roots without tubers. **Stem:** vestiture of 3 types, each sometimes sparse: (1) minute, gland-tipped hairs; (2) long, \pm stiff hairs to 3.5(4) mm; and (3) short hairs < 0.5 mm. **Leaves:** to 28 cm; major leaflets (5)9–19(21), lanceolate or oblanceolate to narrowly elliptic (rhombic), $1-8.5 \times 0.6-2.5$ cm, adaxially minutely pubescent, abaxially with copious gland-tipped hairs and with long hairs 1–3 mm; l:w = 4.0 (3.3–4.8); minor leaflets 15–43, to 2.5 cm, 1–4 pairs between major leaflets. **Inflorescence** minutely glandular, with sparse, short hairs < 0.5 mm and long, stiff, more or less straight or curved hairs to 2 mm. **Fruit** (excluding bristles and sepals) 2.3–

4.0(5.0) \times 2.5–4.0(4.5) mm, glandular, the hypanthium top-shaped to campanulate, shallowly grooved, the ridges, base, and pedicel often with a few scattered, stiff, white hairs to 0.5 mm, the grooves glandular, otherwise glabrous; sepals in fruit 1.5–1.7 mm; bristles 1.0–3.0 mm, in 3–4 rows, lowermost row spreading to ca. 90° or reflexed. $2n = 28$ (Hara and Kurosawa 1968).

Moist or wet soil at swamp or stream edges, grassy areas, meadows, thickets, and roadside ditches; sometimes in dry, open places.

Agrimonia parviflora is widely distributed in Kentucky. The documented county-distribution of the species in the state is as follows: Allen, Bath, Bell, Bourbon, Breckinridge, Butler, Calloway, Campbell, Casey, Clark, Edmonson, Fleming, Floyd, Franklin, Garrard,

Greene, Hardin, Harlan, Jackson, Jefferson, Kenton, Laurel, Lee, Lincoln, Lyon, Madison, Magoffin, Marion, McCracken, McCreary, Meade, Menifee, Montgomery, Morgan, Muhlenberg, Pike, Powell, Pulaski, Rockcastle, Rowan, Trigg, Warren, Whitley, and Wolfe.

A study of the reproductive success and breeding system of *A. parviflora* was recently completed by Brann (1998) (see under Biology: Pollination below).

3. ***Agrimonia pubescens*** Wallroth [Latin *pubis*, downy] Figures 5, 9, 13

Herb 3–16 dm; roots with fusiform tubers. **Stem:** vestiture of 2 or 3 types: (1) minute, gland-tipped hairs, these sparse or sometimes lacking; (2) long, stiff hairs to 3 mm, these often sparse; and (3) short hairs < 0.5 mm; types 2 and 3 not always clearly distinguished. **Leaves:** to 24 cm; major leaflets (3)5–9(13), lanceolate or oblanceolate to oblong or elliptic or sometimes obovate, $1.4\text{--}10.7 \times 2\text{--}4.9$ cm, adaxially sparsely pubescent, abaxially downy-pubescent, the hairs 1–2 mm; l:w = 2.2 (1.7–3.0); minor leaflets 4–11, to 1.7 cm, 1–3 pairs between major leaflets. **Inflorescence** minutely glandular, the glands often sparse or even lacking, with short hairs < 0.5 mm and long, stiff hairs to 2 mm, the long hairs sometimes sparse or even lacking. **Fruit** (excluding bristles and sepals) $4.0\text{--}5.5(6.0) \times 2.3\text{--}4.0$ mm, glandular (sometimes sparsely so), the hypanthium top-shaped to campanulate, shallowly to deeply grooved, the ridges, base, and pedicel with a few scattered, stiff, hairs 0.5–0.8 mm, the grooves often with a strip of conspicuous to inconspicuous white, upwardly appressed hairs < 0.5 mm (rarely seemingly white farinose); sepals in fruit 1.5–1.7 mm; bristles 1.0–3.2 mm, in 3–4 rows, lowermost row spreading to ca. 90°.

Dry to mesic woodlands, woodland edges, disturbed sites.

Agrimonia pubescens is widely distributed in Kentucky. The documented county-distrib-

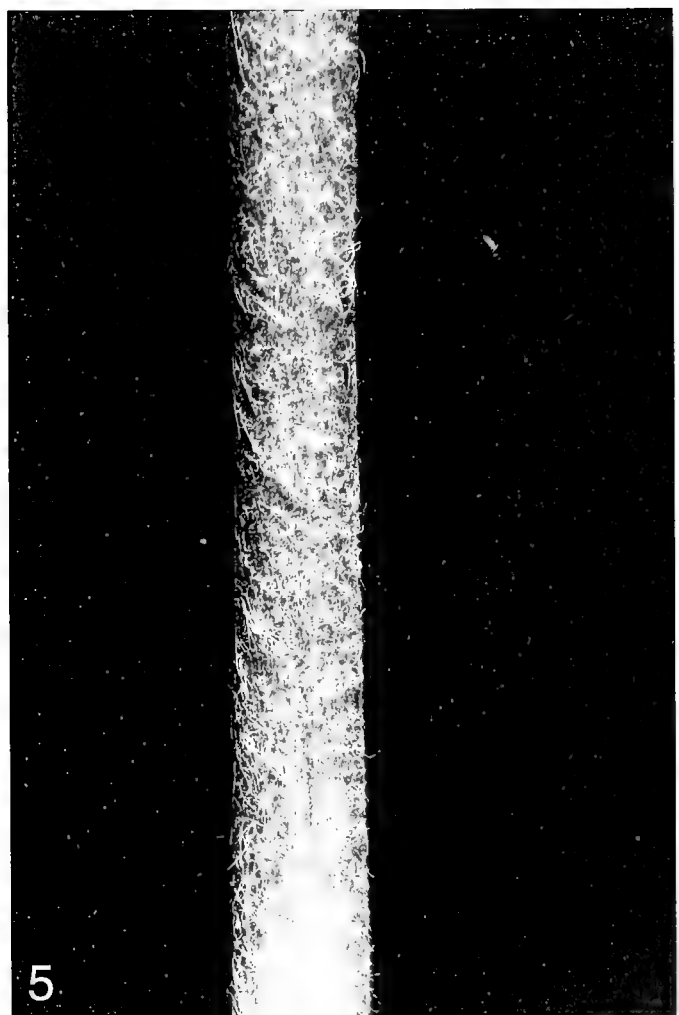
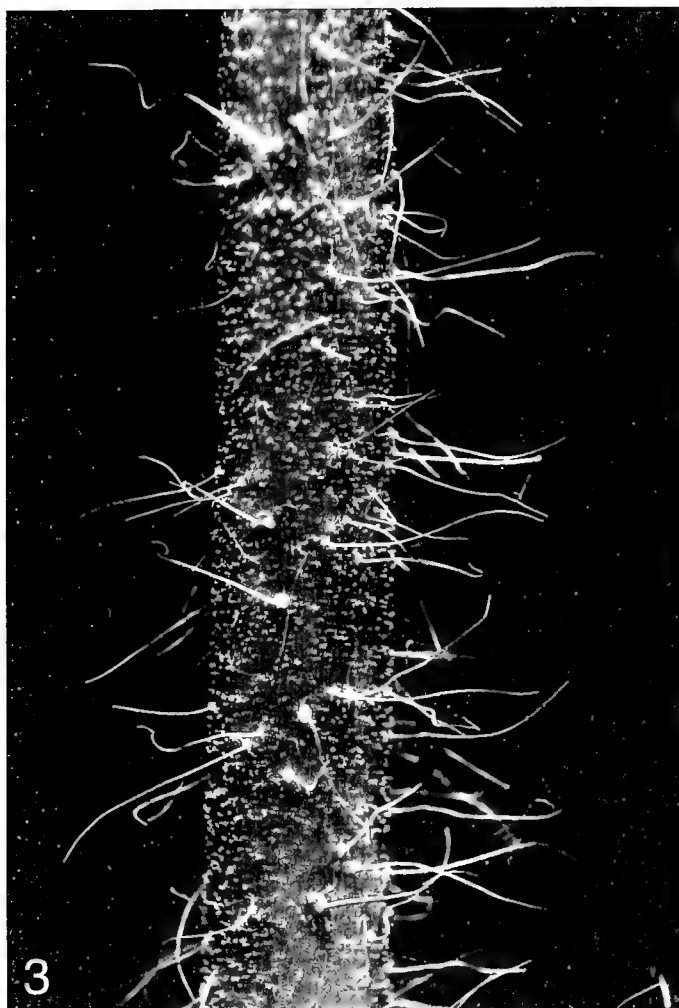
ution of the species in the state is as follows: Anderson, Barren, Boone, Boyle, Bracken, Breathitt, Breckinridge, Bullitt, Caldwell, Campbell, Clark, Clinton, Edmonson, Estill, Fayette, Floyd, Franklin, Gallatin, Green, Greenup, Hardin, Harlan, Hart, Henry, Jackson, Jefferson, Jessamine, LaRue, Laurel, Letcher, Lincoln, Lyon, Madison, Marshall, McLean, McCreary, Meade, Menifee, Muhlenberg, Nelson, Nicholas, Ohio, Oldham, Owen, Pendleton, Pike, Pulaski, Robertson, Todd, Trigg, Warren, Wayne, Wolfe, and Woodford.

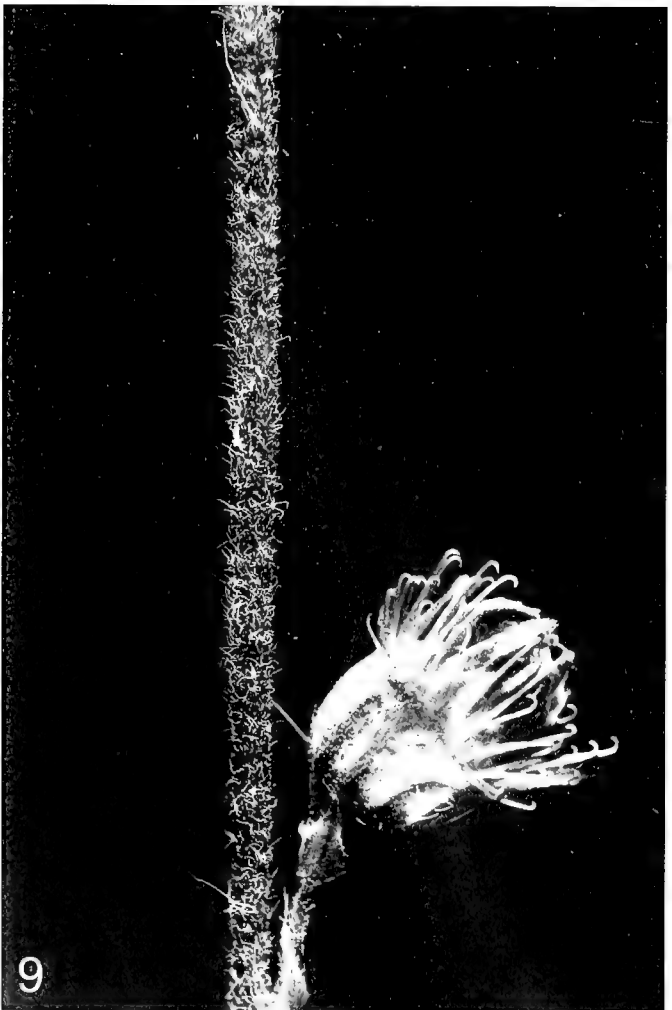
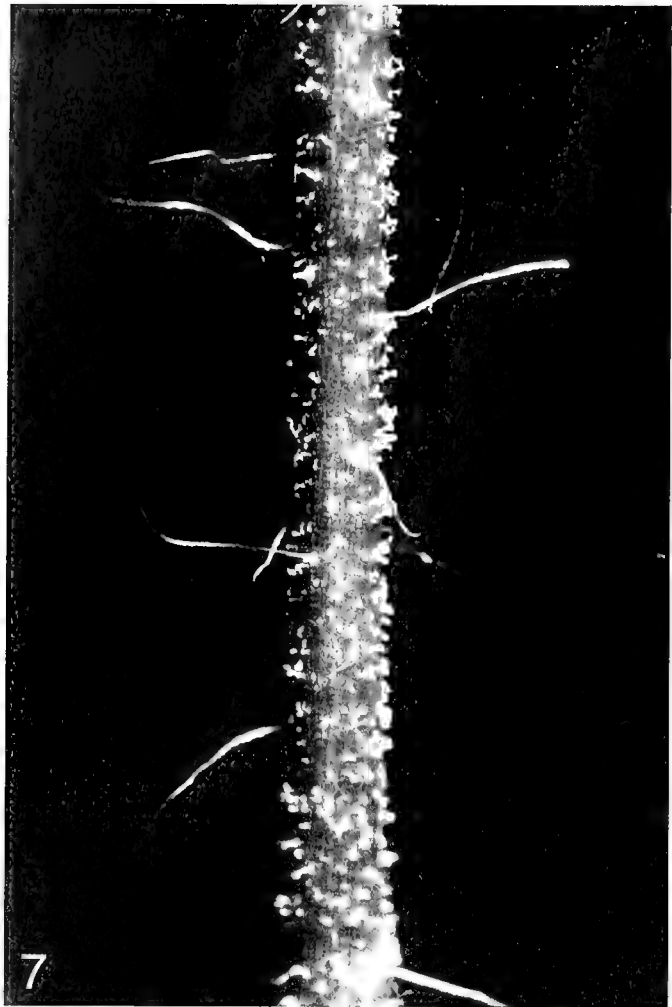
4. ***Agrimonia rostellata*** Wallroth [Latin *rostellum*, beak, alluding to the connivent sepals on the fruit] Figures 6, 10, 14

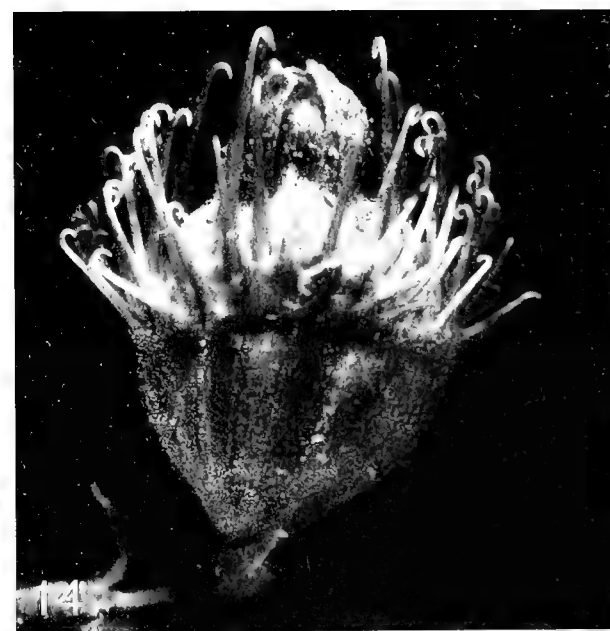
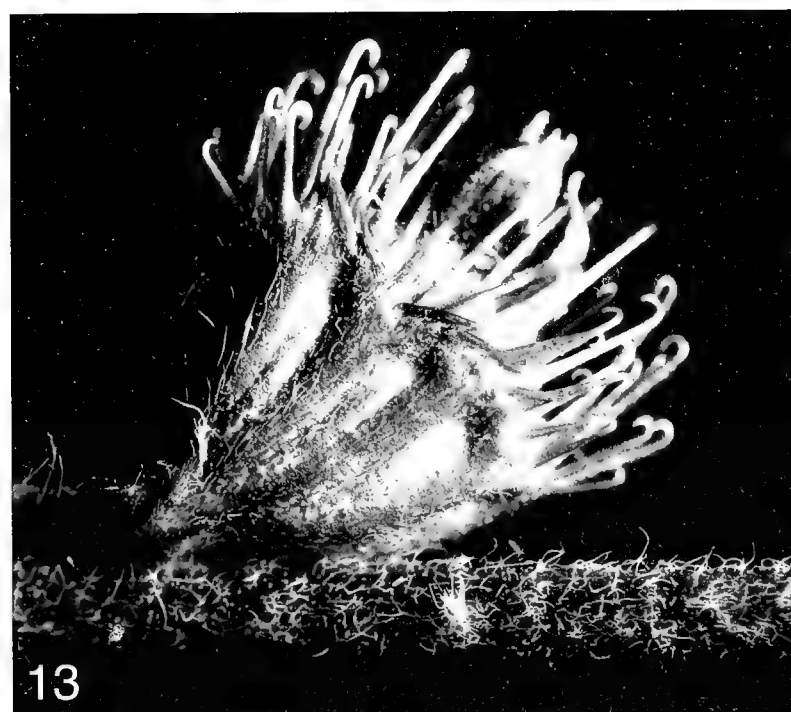
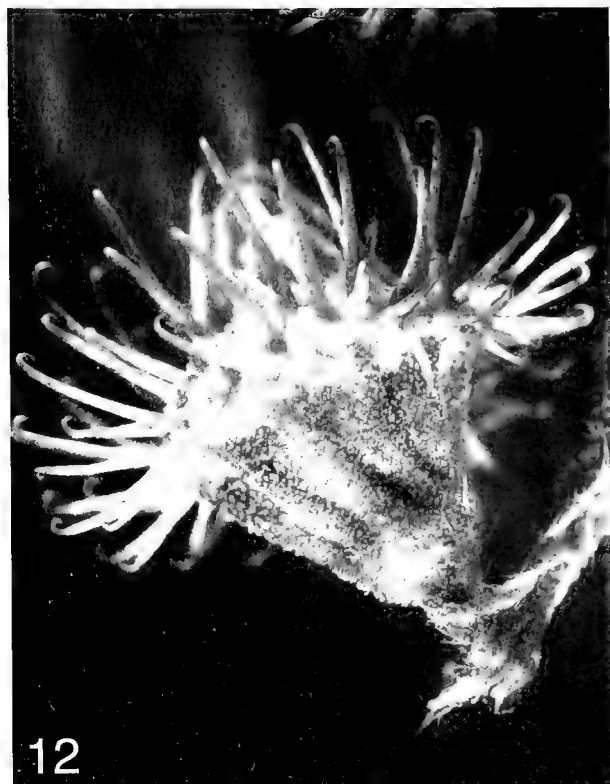
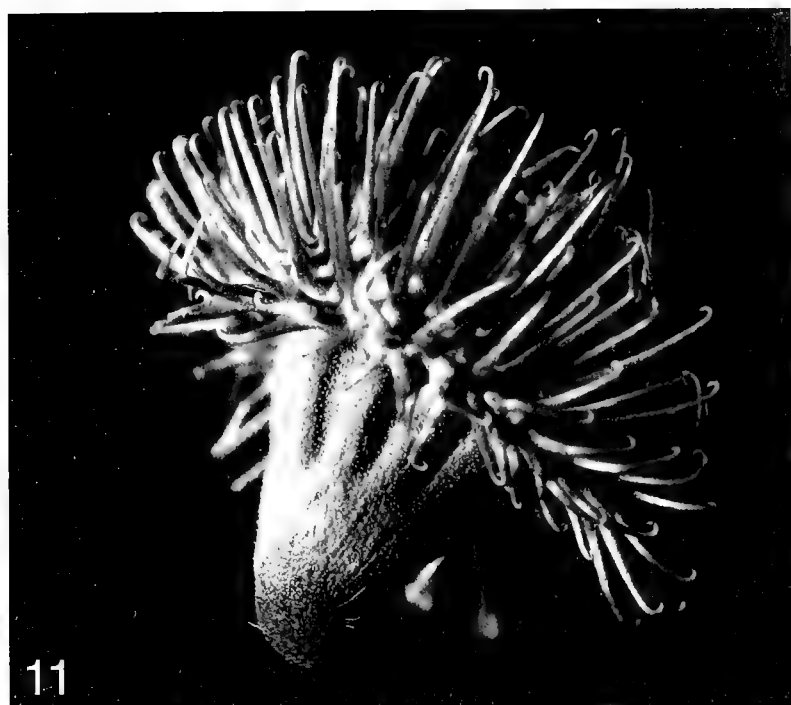
Herb 2–10 dm; roots with fusiform tubers. **Stem:** vestiture of 3 types: (1) minute, gland-tipped hairs, these sparse to abundant; (2) long, stiff hairs to 2.5(3) mm, these sometimes sparse; and (3) sparse, short hairs < 0.5 mm. **Leaves:** to 24 cm; major leaflets 3–9(11), obovate to broadly elliptic, $2.5\text{--}10.5 \times 1.5\text{--}5.6$ cm, adaxially glabrous or sometimes with a few long hairs, abaxially with copious gland-tipped hairs and with long hairs ± 1 mm; l:w = 1.9 (1.6–2.5); minor leaflets 0–8, to 2.5 cm, 1(0–2) pair between major leaflets. **Inflorescence** sparingly to copiously glandular, with sparse, short hairs < 0.5 mm and long, stiff hairs to 1 mm, both types sometimes absent. **Fruit** (excluding bristles and sepals) $3.2\text{--}4.0 \times 2.0\text{--}3.0$ mm, obscurely to conspicuously glandular, otherwise glabrous or sometimes with a few short hairs < 0.2 mm at base or on the pedicel, the hypanthium hemispherical, shallowly grooved; sepals in fruit 1.5–1.8 mm; bristles 1.7–2.0 mm, in 3–4 rows, lowermost row spreading to ca. 90°.

Dry to mesic woodlands, woodland edges, disturbed sites.

Agrimonia rostellata is widely distributed in Kentucky. The documented county-distribution of the species in the state is as follows:







Figures 11–14. *Agrimonia*. “Fruits.” Figure 11, *A. gryposepala* (length 7 mm). Figure 12, *A. parviflora* (length 3 mm). Figure 13, *A. pubescens* (length 4.5 mm). Figure 14, *A. rostellata* (length 3.2 mm).

Allen, Anderson, Ballard, Barren, Bath, Bell, Breathitt, Breckinridge, Bullitt, Caldwell, Calloway, Campbell, Carlisle, Carter, Clark, Clinton, Crittenden, Daviess, Edmonson, Estill, Fayette, Floyd, Garrard, Grant, Greenup,

Hardin, Hart, Hickman, Jefferson, Jessamine, Kenton, LaRue, Laurel, Lawrence, Letcher, Lewis, Lyon, Madison, McCreary, McLean, Meade, Menifee, Morgan, Nelson, Nicholas, Oldham, Pike, Powell, Pulaski, Robertson,

←

Figures 7–10. *Agrimonia*. Median portions of inflorescence axis, showing vestiture. Figure 7, *A. gryposepala* (axis diam 1.5 mm). Figure 8, *A. parviflora* (axis diam. 2 mm). Figure 9, *A. pubescens* (axis diam. 2 mm). Figure 10, *A. rostellata* (axis diam. 1 mm).

Rockcastle, Rowan, Spencer, Taylor, Todd, Trigg, Warren, Wayne, and Woodford.

EXCLUDED SPECIES

Agrimonia eupatoria L.

Agrimonia eupatoria has been ascribed to Kentucky several times starting with M'Murtrie (1819), but no documenting Kentucky specimens are known to us. The species may not even be naturalized in North America (Kline and Sørensen 1990) in spite of the many reports that it is so. It was, however, recorded in 1924 as a wool-waste plant from Massachusetts (Weatherby 1924), which may explain, in part, Fernald's (1950) attributing it to "waste places and old fields, local, Mass., Wisc. and Minn." (We have seen century-old Minnesota specimens at MU labelled as *A. eupatoria*; they are *A. gryposepala*.) Gleason and Cronquist (1991) wrote merely that the species is "occasionally intr. In our range." Kartesz and Meacham (1999) ascribed the species, in eastern North America, to three Canadian provinces and eight U.S. states (but not Kentucky); and, in the west, to one province and two states; most of these reports are based on literature records, not on first-hand study of specimens. But Kline and Sørensen (n.d.), in their account of *Agrimonia* for *Flora of North America*, excluded the taxon from the FNA area: "*Agrimonia eupatoria*, a European species, has been sporadically cultivated in the flora area. We can find no evidence that this introduced species, which rarely escapes cultivation, has become an established element of the flora." Earlier, Bicknell (1896) reached the same conclusion: "... the true *Agrimonia Eupatoria* is not known at all as an American plant. . . ."

Of *A. eupatoria* Weatherby (1924) wrote that "it resembles our native *A. gryposepala* in that the stem and the rachis of the inflorescence are clothed with minute glandular puberulence mixed with long, non-glandular hairs. It is readily recognized, however, by its commonly more compact habit, the lower internodes of the stem tending to be short, thus bringing the leaves close together, by its generally smaller leaflets, and by the characters of the fruiting calyx. The body of the mature hypanthium is rather narrowly top-shaped and measures from the base to the point of inser-

tion of the hooked bristles about 5 mm. In *A. gryposepala* the corresponding measurement is about 3 mm."

With respect to Kentucky, the species is one that could have been—or is—cultivated here in medicinal plant or other gardens. But we know of no evidence for such cultivation. (Seeds said to be of *A. eupatoria* are available from several websites.)

In 19th century U.S. floristic literature the accounts of *Agrimonia* were hopelessly confused as to both nomenclature and taxonomy (Bush 1916), with "*A. eupatoria*" being often a catch-all name for any U.S. agrimony except the distinct *A. parviflora*. Clarification of the nomenclature and taxonomy of the U.S. representatives of the genus began in the last years of the 19th century and the early years of the 20th century (Bicknell 1896, 1901a, 1901b; Britton and Brown 1897; Robinson 1900, 1901; Robinson and Fernald 1908; Rydberg 1913). Some authors, though, are still somewhat confused; we hide their identity to protect the guilty.

The 19th century confusion in *Agrimonia* is best illustrated by the accounts of the genus published in the 1890s in two major floristic works for northeastern U.S. The sixth edition of Gray's *Manual* (Gray 1890) includes only two species of the genus—*A. eupatoria* and *A. parviflora*. The first edition of the Britton and Brown *Illustrated flora* (Britton and Brown 1897) includes six species, excluding *A. eupatoria* and commenting that "The European *A. Eupatoria* L. differs markedly in foliage and fruit from any of our species." Following the lead of the *Illustrated flora*, the 7th edition of Gray's *manual* (Robinson and Fernald 1908) included six species but not *A. eupatoria*. The most recent edition of the *Manual* (Fernald 1950) includes seven, *A. eupatoria* having been reinstated—probably in error—as a member of the flora.

Agrimonia microcarpa Wallr.

This species has been ascribed to Kentucky in several works (e.g., Browne and Athey 1991; Fernald 1950; Greenwell 1935; Kearney 1893; Medley 1993; Radford et al. 1968). Even a county of occurrence—Bullitt—has been mentioned. We made an unsuccessful attempt to locate confirming specimens. According to Kline and Sørensen (1990), this

southeastern species ranges north to North Carolina, Tennessee, and Missouri.

Agrimonia microcarpa would key to *A. pubescens* in the key above. Although recognized as a distinct taxon by various authors, its distinguishing characters seem to us to be rather weak, overlapping in most cases with those of *A. pubescens*; it has been considered a variety of that species (Ahles 1964; Radford et al. 1968). To separate *A. microcarpa* from *A. pubescens* the following features have been among those used: stem hairs 3–4 mm (*m*) vs. 1–3 mm (*p*); major leaflets 3–7 (*m*) vs. 3–13 (*p*); major leaflets broadly rounded distally (*m*) vs. acute to obtuse (*p*); stipules \pm falcate to \pm half round and deeply incised (*m*) vs. \pm broadly half ovate, incised but not deeply (*p*); and mature hypanthium with a distinct rim and as broad as long (*m*) vs. mature hypanthium with a obsolete rim and usually longer than broad (*p*).

Agrimonia striata Michx.

Agrimonia striata is a northern and western species ranging south, in eastern U.S., to Iowa, Illinois, Michigan, Ohio, West Virginia, Pennsylvania, and New Jersey (Kline and Sørensen 1995). Its roots are slender (non-tuberous); it has (3)5–9(11) major leaflets; its abaxial leaflet surfaces are sparingly pubescent, the hairs to 1 mm and confined mostly to veins; its inflorescence is glandular and pubescent, the hairs 1–2 mm; and its hypanthium grooves have a strip of minute, appressed hairs. Voss (1985) pointed out that the stipules of its mid-cauline leaves usually have a prolonged lanceolate terminal tooth or lobe.

BIOLOGY

Biological data on *Agrimonia*, at least on our North American species, are relatively scarce. The best sources of such data on the European species known to us are the accounts of *Agrimonia* in two editions of Hegi (1923, 1995). Only *A. eupatoria*, long used medicinally in Europe and elsewhere, has been studied with any degree of thoroughness.

Pollination

Few observations on pollination of agrimonies are available. The species, at least in Europe, bears pollen flowers, which do not produce nectar and are visited by pollen-eating

insects. “Flies and bees are attracted to the slender spikes of flowers [of *A. eupatoria*] by a scent reminiscent of apricots” (RDA 1981).

The pollination mechanism of *A. eupatoria* was described thus by Knuth (1908):

At the base of the two styles in this species there is a fleshy ring that looks like a nectary, though no secretion has been observed. The 5–7 stamens on the margin of this disk attain the same level as the stigmas, and their anthers dehisce laterally. The anthers incline inwards, and therefore come into contact with the stigmas. The individual flowers bloom for a single day only, and open very early in the morning. The stamens, which are at first divergent, bend inwards in the course of the day, until they touch one another and the stigmas. Comparatively few insects visit the flowers, but these may bring about either cross- or self-pollination. From the above description it is clear that the latter occurs automatically, and it is obviously effective.

Palynological data on *Agrimonia* (i.e., *A. eupatoria*) are included in Hegi (1995; light microscope and SEM photos of pollen); Erdtman (1952; description of pollen); and Reitsma (1966; line drawings of pollen).

The pollen of *A. eupatoria* was described thus by Erdtman (1952): “3-colpor(oid)ate (constricticopate), prolate ($48 \times 31 \mu$). Sexine thicker than nexine, tegillate; endosexine considerably thinner than ectosexine. The latter finely striate.”

We include, as Figure 15, a photomicrograph of pollen of *A. parviflora*.

Mueller (1883) recorded as pollinators of European *A. eupatoria* the following: Diptera–Syrphidae, nine species representing six genera (*Ascia*, *Eristalis*, *Melanostoma*, *Melithreptus*, *Rhyngia*, *Syritta*); Muscidae, one species (*Anthomyia*); and Hymenoptera–Apidae (a “small” species of *Halictus*). To these Knuth (1908) added a species of *Syrphus* (Diptera: Syrphidae) and a species of *Apis*, one of *Bombus*, and one of *Prosopis* (Hymenoptera: Apidae). Clapham et al. (1987) mentioned “Diptera and Hymenoptera.”

North American data on pollination of agrimonies are even fewer than those from Europe. Indeed, we have found only two reports: C. Robertson (1928), working near Carlinville, Illinois, recorded a species of *Chloralictus* (Hymenoptera: Halictidae) as a visitor to *A. striata*; and Macroberts and Macroberts (1997) wrote that “captured pollinators [of *A.*

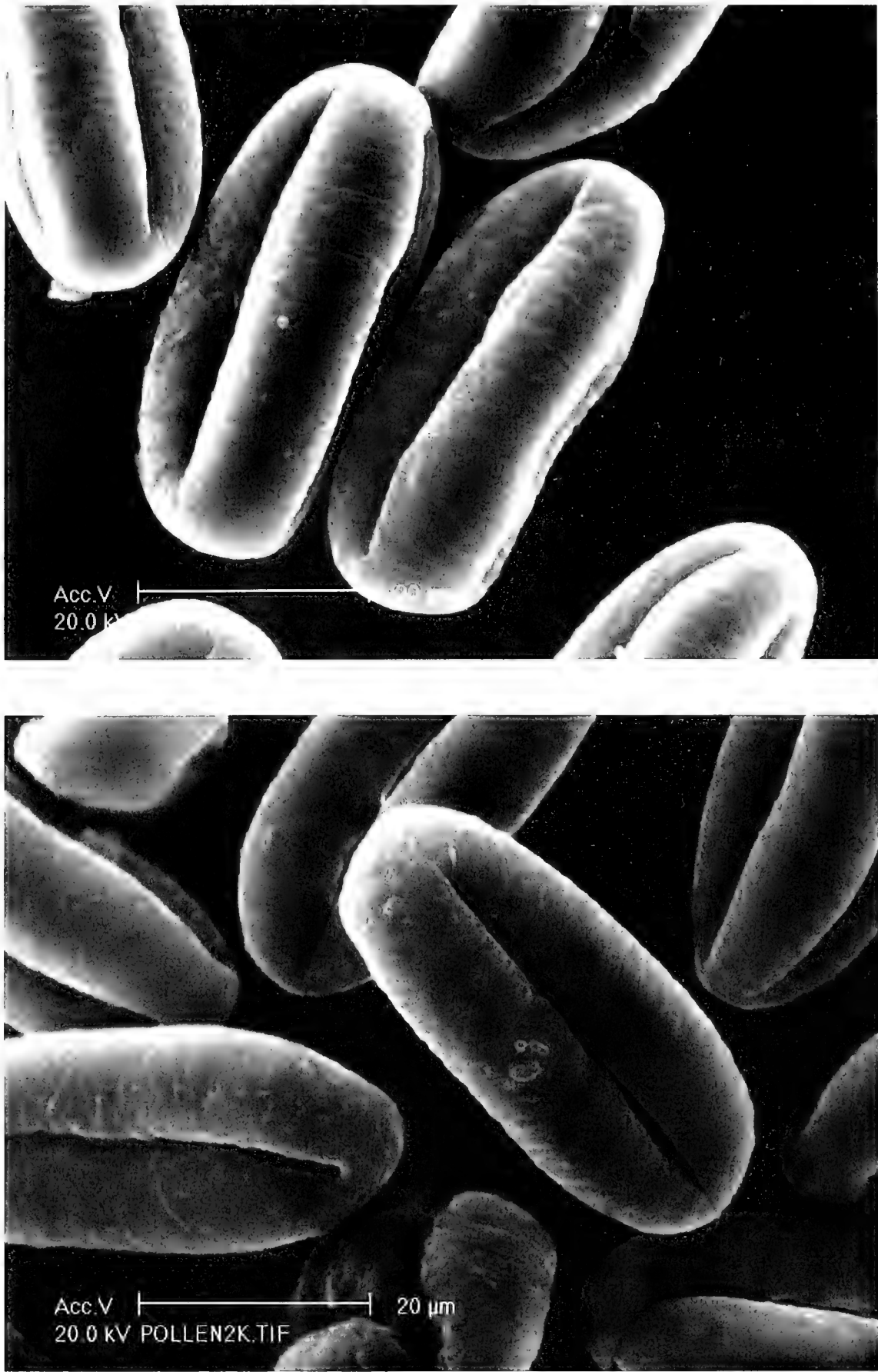


Figure 15. *Agrimonia parviflora*. Pollen grains, $\times 2000$. (Voucher: *Thieret* 52394, Rowan County, Kentucky, 29 Aug 1980 [KNK]). Photomicrograph by Brenda K. Racke.

incisa, a southeastern U.S. species] were small bees of the subfamily Halictinae (Hymenoptera, Apoidea, Halictidae), all probably the same species.”

In a study of the breeding system of *A. parviflora*, Brann (1998) found the species to be self-compatible. Its flowers opened by 0900 and closed by 1400 and were open for only 1 day. Once a flower opened, the availability of its pollen for dispersal and the receptivity of its stigmas were synchronous.

Dispersal

The fruits of *Agrimonia*, with their hooked bristles, are dispersed by animals, including humans, to the fur or clothing of which they readily become attached (Coffey 1993; Keville 1991; Macroberts and Macroberts 1997; RDA 1981; Ridley 1930; S.M. Robertson 1973). Dispersal even by birds on rare occasions might be possible as suggested by the following quote from Swink and Wilhelm (1994): “The senior author once rescued a Goldfinch that was hopelessly trapped in the prickly fruits of [*A. gryposepala*].” Macroberts and Macroberts (1997) suggested that many of the fruits, though seemingly well adapted for long-range dispersal, probably simply drop near the parent stem. Such in-situ dispersal, it seems to us, would result in close groups of plants, but—except in one instance, a group of ca. 20 individuals of *A. parviflora*—the agrimonies we have seen in Kentucky occur as scattered plants.

Dispersal by water, at least for streamside plants, is presumably possible, the fruits being able to float for about a week, at least in the case of *A. eupatoria* (Ridley 1930).

Tuberous Roots

Half of the North American species of *Agrimonia* (*A. incisa*, *A. microcarpa*, *A. pubescens*, and *A. rostellata*) have tuberous roots the function of which is unknown. Macroberts and Macroberts (1997) suggested that “all of these species might occur in fire-dependent or droughty areas where food reserves or the alternatives of clonal reproduction might be important.” Perhaps water storage is important, too: Our Kentucky species, except often *A. parviflora*, grow in dry—sometimes impressively dry—sites.

Cytology

The base chromosome number of *Agrimonia* is $x = 7$ (Darlington and Wylie 1955; Hegi 1995; Iwatsubo et al. 1993). The U.S. species with reported counts are *A. parviflora*, a tetraploid ($2n = 28$; Hara and Kurosawa 1968), *A. gryposepala*, an octoploid ($2n = 56$; Brittan 1953), and *A. striata*, a tetraploid or octoploid ($2n = 28, 56$; Hegi 1995). *Agrimonia* in Japan includes species at the $4\times$, $6\times$, and $8\times$ levels. In Eurasia, *A. eupatoria* is a tetraploid ($2n = 28$) and *A. pilosa* is an octoploid (Hegi 1995; Löve and Löve 1961). A hybrid, *A. eupatoria* \times *A. procera*, is a hexaploid ($2n = 42$) as is *A. nipponica* \times *A. pilosa* var. *japonica* (Iwatsubo et al. 1993).

MEDICINAL

The best-known species of *Agrimonia* is the Eurasian *A. eupatoria*. In older herbal literature it was almost panacean, being used in a number of ways for an impressive array of ills from A (asthma) almost to Z (warts) (Hegi 1995). In recent literature (e.g., Bartram 1995; Bown 1995; Duke 1985, 1997; Foster and Duke 2000; Keville 1991; Ody 1993; Schauenberg and Paris 1977; Swanston-Flatt et al. 1990; Wood 1997) it is said to have been used to treat a diversity of ailments including amenorrhea, bed-wetting, conjunctivitis, cystitis, diabetes, diarrhea, eczema, gallstones, gout, hemorrhoids, incontinence, laryngitis, migraines, nosebleed, rheumatism, skin inflammation, sore throats, and ulcers. A somewhat more restrained assessment of the virtues of the species (LRNP 1995) concludes that “it does appear to have justifiable use as a mild antiseptic and topical astringent” but cautions that “internal uses of this herb require further verification.” Commission E approves *A. eupatoria* to treat diarrhea, inflamed mucous membranes of the mouth and throat, and mild, superficial inflammation of the skin (Blumenthal 1998). The astringent properties of *A. eupatoria* presumably derive from its high tannin content: roots, 25.8%; rhizome, 16.4%; leaves, 16%; and stem, 5.8% (Hegi 1995).

Wood’s (1997) 27-page discussion of the medicinal and other virtues of *Agrimonia* is the most detailed and wide-ranging that we have seen.

A tea made from *A. eupatoria* has been

used to treat dysentery (Anonymous 1856). The plant, in a mixture of powdered frogs and human blood, was once “recommended for all internal hemorrhages” (Johnson 1862). Hartwell (1982), in a survey of plants used against cancer, included many data from historical literature on *A. eupatoria*.

Langer and Kubelka (1998) described rhizome anatomy of *A. eupatoria* in comparison to that of *Potentilla erecta* (Radix Tormentillae), another medicinal plant.

Within the past 2 decades or so, *A. pilosa*, another Eurasian species, has been increasingly written about, e.g., the effect of a water extract of the plant on tumors (Miyamoto et al. 1987) and acute pulmonary thrombosis (Hsu et al. 1987). In Chinese herbal medicine *A. pilosa* is used to expel tapeworms (Nigg and Seigler 1992, and “Agrimony” is one of the herbs that “regulate” the blood (Ehling and Swart 1996).

The mention of *A. eupatoria* in older literature on North American medicinal plants may or may not refer to this European species, which is quite similar to some of our indigenous taxa of the genus. The plant may well have been introduced early to the continent as a medicinal plant (S.M. Robertson 1973), but problems with identification remain. Erichsen-Brown (1989) referred these early records unequivocally to the native *A. striata*. Why she chose this species and not any of the several other indigenous ones is not explained; her selection is another unverifiable datum.

Agrimonia was early noted in reports of the uses of American medicinal plants by Europeans. In the “first systematic publication concerning the American materia medica” (Lloyd in Smith 1812), that of Johann David Schoepf (1787), *A. eupatoria* is described as having astringent, roborant, prophylactic, diuretic, and vulnerary properties; the name *A. eupatoria* is probably a misidentification for one of the North American taxa. Cutler (1785), in his work on some of the “vegetable productions” of New England, wrote of *Agrimonia* (no specific epithet given but presumably a native species) as growing “by fences” and being useful to treat fevers and jaundice. Smith (1812), in his “dispensatory” on Ohio Valley plants, described “agrimoney” (again no species indicated) as “a native of the woods [and thus certainly an indigenous species], but friendly

to cultivation.” Its virtues included being useful as a tonic and in diabetes, involuntary emission of urine, and dysentery and “other fluxes.” Rafinesque (1828) called *A. eupatoria* a “mild astringent, tonic, and corroborant” and noted its use for diarrhea, dysentery, “relaxed bowels,” and asthma. He included a color illustration in his account of the species, but this is unidentifiable to species. His description of the range and habitat of the plant—“The *Agrimonia Eupatoria* is spread from Canada to Missouri and Carolina, and grows in woods, fields, glades and near streams”—is obviously based on an indigenous species. In several Shaker communities (1847–1874) *A. eupatoria* was “highly recommended in bowel complaints, gravel, asthma, coughs, and gonorrhea.” Again, that this “*A. Eupatoria*” was a native U.S. species is made almost certain by the statement that it was “found by the roadsides and borders of fields, Can. and U.S.” (Miller 1976).

Eastern and central North American Native Americans included at least two species of *Agrimonia*—*A. gryposepala* and *A. parviflora*—among the plants they used medicinally (Erichsen-Brown 1989; Moerman 1998). Snakebite, jaundice, diabetes, nosebleed, urinary troubles, nephritis, and diarrhea were among the problems for which the agrimonies were taken; they were also used—conflictingly, it seems to us—as an antidiarrheal medication and as an emetic, though probably not simultaneously.

“Agrimony” (presumably *A. eupatoria*) is one of the the “Twelve Healers” divinely revealed to the British physician Edward Bach, M.D. (Harrar and O'Donnell 1999). Bach wrote of this herb in his system of healing with flower essences: “An herb . . . for cheerful, peaceful people who loathe discord and whose jovial exterior covers inner torment, sometimes resulting in problems with drugs or alcohol.”

Various “Agrimony” websites list sources of seeds (*A. eupatoria*, *A. pilosa*) and medicinal preparations and uses including even a homeopathic remedy from *A. eupatoria*.

MISCELLANEOUS

Although the species of *Agrimonia*, as we know them in Kentucky, are anything but “weedy,” at least *A. gryposepala* can, on oc-

casion, be troublesome. Of this species Muenscher (1955) wrote: "badly infested fields should be plowed and planted to a cultivated crop for a season." A color illustration of this species is given in Alexander (1932). Drawings of the seedling stages of *A. gryposepala* are given in Kummer (1951).

Agrimonia eupatoria was even used as a sleep aid, as the following Old English verse testifies (Addison 1985):

If it be leyd under a mann's head
He shall sleepyn as he were dead.
He shall never drede ne wakyn,
Till fro under his head it be taken.

Almost certainly any of our North American species would be as soporifically efficacious.

Anti-ophidian properties were even ascribed to *A. eupatoria* (Law 1973). The plant was used in a charm to ward off snakes. One of its old English names was sticklewort, hence this bit of doggerel:

He that hath sticklewort by
Knows no snake shall draw him nigh.

Agrimonia eupatoria was implicated by O'Donovan (1942) in the production of phytophotodermatitis in humans, but he established a probable connection between plant and patient in only one of 14 cases. Mitchell and Rook (1979) suggested that irritation (from the plant's trichomes?) rather than photosensitization was the cause of the condition. Later research showed that an alcoholic extract of *A. eupatoria* has "a very slight photosensitizing action" (Dijk 1963). Dijk and Berrens (1964) "assumed" but did not demonstrate that "Agrimonias contain[s] photosensitizing furocoumarins" as do other, much more potent photosensitizing plants, e.g., *Ammi majus*, *Pastinaca sativa*, and *Ruta graveolens*. Obviously more study of the problem is needed.

A tea can be made by steeping leaves and stems of *A. parviflora* and *A. rostellata* in boiling water, cooling, and serving with sugar or lemon (Cheatham and Johnston 1995). A similar tea, from *A. eupatoria*, is used in Britain as a "purifier of the blood" (Hulme 1912) and is said to be "particularly adapted to people who live poorly, and imperfectly digest their bad food" (Anonymous 1856).

Agrimonia eupatoria (leaves and stems) is a

minor dye plant, giving a stable yellow, gold, or orange depending on the mordant (Addison 1985; BBG 1984; Hutchinson 1972; S.M. Robertson 1973; Keville 1991; Lushchevskaya 1937; Usher 1974) and on the month of harvest (Johnson 1862). Our native species would probably be similarly useful.

Agrimonia eupatoria "contains tannin, and has been used in dressing leather" (Pratt 1905). Flowers of the species, with their "honey flavour," were once added to mead; the dried plant, with its fragrance, was included in "sweet sachets and pot-pourri" (Huxley 1992).

Several abnormalities have been noted in the development of flowers/inflorescences of *Agrimonia*, the reports all European (and thus the plant being probably *A. eupatoria*). Masters (1869) wrote that the genus is among those in which suppression of the androecium occurs most frequently and that leaves may develop in the center of a flower. Anomalies in the number of sepals, the position of bracts, and the number of bractlets are known (Phelouzat 1963). Fasciation in the inflorescence results in a compact, terminal mass of flowers, the whole being somewhat reminiscent of a head of a member of the Asteraceae (Phelouzat 1963; Schimper 1854). Fusion of two petals can form a structure similar to the keel of a legume flower (Moquin-Tandon 1842).

The petals of *A. eupatoria*, normally yellow, can on rare occasions, be white. Seeds of this white-flowered form, when planted, "bred true to white flower-colour" (Nelmes 1929).

Of the origin of the name *Agrimonia* we may choose from the four possibilities listed by Quattrocchi (2000): "Possibly from the Greek *argemone*, *argemon*, ancient name used by Dioscorides, Plinius and Galenus for the poppy; or from *argemonion* ancient Greek name applied by Dioscorides to the anemone; or from *agros* "field" and *monos* "alone, lonely"; or from *agrios*, *agrimaios* (agra) "wild." Another source (HcVN 2000) derived "agrimony" from a "Greek word describing plants which healed the eyes."

ACKNOWLEDGMENTS

We thank the Lloyd Library, Cincinnati, Ohio, for literature; Dr. David M. Brandenburg, Dr. Jerry H. Carpenter, Dr. David Hewitt, Dr. Miriam S. Kannan, Dr. Genevieve J. Kline, Brenda K. Racke, Annette D. Skinner,

Dr. Paul Sørensen, Dr. Ralph Thompson, and Dr. Michael A. Vincent for aid; Dr. Elsa M. Zardini for permission to reproduce her drawings of *A. parviflora* as Figures 1 and 2; and the curators of the following herbaria for loans of specimens: APSC, BERE, Cumberland College, DHL, EKY, KSNPC, KNK, KY, MDKY, MOR, MU, NCU, PH, WKU, and private herbarium of Randy Seymour, Upton, Kentucky (repeated requests [by phone, FAX, and e-mail] for loan of specimens from Murray State University went unacknowledged).

LITERATURE CITED

- Addison, J. 1985. The illustrated plant lore. Sidgwick and Jackson, London, U.K.
- Ahles, H. E. 1964. New combinations for some vascular plants of southeastern United States. *J. Elisha Mitchell Sci. Soc.* 80:172–173.
- Alexander, E. J. 1932. *Agrimonia gryposepala*. *Addisonia* 17:19–20, plate 554.
- Anonymous. 1856. Agrimony tea. *Gard. Chron.* 1856:584.
- Bartram, T. 1995. Encyclopedia of herbal medicine. Grace Publishers, Christchurch, Dorset, U.K.
- [BBG] Brooklyn Botanic Garden. 1984. Handbook on dye plants and dyeing. *Brooklyn Bot. Gard. Rec.* 20(3).
- Bicknell, E. P. 1896. The North American species of *Agrimonia*. *Bull. Torrey Botanical Club* 23:508–523.
- Bicknell, E. P. 1901a. The nomenclature of the New England agrimonies. *Bull. Torrey Bot. Club* 28:102–108.
- Bicknell, E. P. 1901b. Still further notes on the agrimonies. *Bull. Torrey Bot. Club* 28:514–518.
- Blumenthal, M., senior ed. 1998. The complete Commission E monographs. American Botanical Council, Austin, TX; Integrative Medicine Communications, Boston, MA.
- Bown, D. 1995. Encyclopedia of herbs and their uses. Dorling Kindersley, London, U.K.
- Brann, M. 1998. The reproductive success and breeding system of *Agrimonia parviflora* Aiton under different environmental conditions. M.S. Thesis. Truman State Univ., Kirksville, MO.
- Braun, E. L. 1943. An annotated catalog of spermatophytes of Kentucky. Published by the author, Cincinnati, OH.
- Brittan, N. H. 1953. Cytotaxonomy of some species of *Agrimonia* L. *Proc. Seventh Internat. Bot. Congr.* [1950]: 278.
- Britton, N. L., and A. Brown. 1897. An illustrated flora of the northern United States, Canada and the British Possessions. Volume II: Portulacaceae to Menyanthaceae. Charles Scribner's Sons, New York, NY.
- Browne, E. T., Jr., and R. Athey. 1992. Vascular plants of Kentucky. Univ. Press of Kentucky, Lexington, KY.
- Bush, B. F. 1916. The Missouri agrimonies. *Ann. Missouri Bot. Gard.* 3:309–318.
- Cheatham, S., and M. C. Johnston. 1995. The useful wild plants of Texas. Useful wild plants, Austin, TX.
- Clapham, A. R., T. G. Tutin, and D. M. Moore. 1987. Flora of the British Isles. 3rd ed. Cambridge University Press, Cambridge, U.K.
- Coffey, T. 1993. The history and folklore of North American wildflowers. Facts on File, New York, NY.
- Cutler, M. 1785. An account of some of the vegetable productions naturally growing in this part of America [i.e., New England]. Reprint, with biography of Manasseh Cutler by J.U. Lloyd, as *Bull. Lloyd Libr.* 7 (Reproduction Ser. 4). 1903.
- Darlington, C. D., and A. P. Wylie. 1955. Chromosome atlas of flowering plants. George Allen and Unwin, London, U.K.
- Dijk, E. van. 1963. Phytophotodermatitis. *Excerpta Med.*, Sect. 13, *Dermatol. Venereol.* 18:371–373.
- Dijk, E., and L. Berrens. 1964. Plants as an ecological factor in phytophotodermatitis. *Dermatologica* 129: 321–328.
- Duke, J. A. 1985. CRC handbook of medicinal herbs. CRC Press, Boca Raton, FL.
- Duke, J. A. 1997. The green pharmacy. Rodale Press, Emmaus, PA.
- Ehling, D., and S. Swart. 1996. The Chinese herbalist's handbook. InWord Press, Santa Fe, NM.
- Erdtman, G. 1952. Pollen morphology and plant taxonomy. Angiosperms. *Chronica Botanica*, Waltham, MA.
- Erichsen-Brown, C. 1989. Medicinal and other uses of North American plants. Dover Publications, New York, NY. Reprint of a work first published in 1979.
- Fernald, M. L. 1950. Gray's manual of botany. American Book Co., New York, NY.
- Foster, S., and J. A. Duke. 2000. A field guide to medicinal plants and herbs of eastern and central North America. Houghton Mifflin Company, New York, NY.
- Gleason, H. A., and A. Cronquist. 1991. Manual of vascular plants of northeastern United States and adjacent Canada. New York Botanical Garden, New York, NY.
- Gray, A. 1890. Manual of the botany of the northern United States. 6th ed., revised by S. Watson and J.M. Coulter. Ivison, Blakeman, and Company, New York, NY.
- Greenwell, R. A. 1935. A flora of Nelson County, Kentucky. Nazareth College, Louisville, KY.
- Hara, H., and S. Kurosawa. 1968. Cytotaxonomical notes on some Asiatic species of *Agrimonia*. *J. Jap. Bot.* 43: 392–400.
- Harrar, S., and S. A. O'Donnell. 1999. The woman's book of healing herbs. Rodale Press, Emmaus, PA.
- Hartwell, J. L. 1982. Plants used against cancer. Quarterman Publications, Lawrence, MA.
- (HcVN) HcVN Health and Healing Tips. 2000. *Agrimonia eupatoria*. Website (hcvn.com) accessed 15 Aug 2000.
- Hegi, G. 1923. Illustrierte Flora von Mittel-Europa. Volume 4(2). A. Pichler's Witwe & Sohn, Wien, Austria.
- Hegi, G. 1995. Illustrierte Flora von Mittel-Europa. Volume 4(2A). 3rd ed. Blackwell Wissenschafts-Verlag, Berlin, Germany.

- Hsu, M. F., J. H. Young, J. P. Wang, and C. M. Teng. 1987. Effect of hsien-ho-t'sao (*Agrimonia pilosa*) on experimental thrombosis in mice. *Am. J. Chin. Med.* 15: 43–51. [Original not seen; abstract in Medline (su: Agrimonia, 88074110), accessed 6 Jun 2000.]
- Hulme, F. E. 1912. Familiar wild flowers. Cassell and Company, London, U.K.
- Hutchinson, J. 1972. British wild flowers. Volume 1. David and Charles, Newton Abbot, U.K.
- Huxley, A., editor-in-chief. 1992. The new Royal Horticultural Society dictionary of gardening. Macmillan, London, U.K.
- Iwatsubo, Y., M. Mishima, and N. Naruhashi. 1993. Chromosome studies of Japanese *Agrimonia* (Rosaceae). *Cytologia* 58:453–461.
- Jain, D. K., and V. Singh. 1973. Structure and development of hairs in *Agrimonia eupatorium* [sic] L. *Curr. Sci.* 42:434–436.
- Johnson, C. P. 1862. The useful plants of Great Britain. William Kent, London, U.K.
- Kartesz, J. T., and C. A. Meacham. 1999. Synthesis of the North American flora. Version 1.0. CD-ROM. North Carolina Botanical Garden, Univ. North Carolina, Chapel Hill, NC.
- Kearney, T. H., Jr. 1893. Notes on the flora of southeastern Kentucky, with a list of plants collected in Harlan and Bell counties in 1893. *Bull. Torrey Bot. Club* 20: 474–485.
- Keville, K. 1991. The illustrated herb encyclopedia. Mallard Press, New York, NY.
- Kline, G. J., and P. D. Sørensen. 1990. Lectotypification and synonymy of *Agrimonia gryposepala* Wallroth (Rosaceae). *Taxon* 39:512–515.
- Kline, G. J., and P. D. Sørensen. 2000. The genus *Agrimonia* (Rosaceae) in Illinois. *Erigenia* 18:13–19.
- Kline, G. J., and P. D. Sørensen. n.d. *Agrimonia*. Account prepared for and submitted to *Flora of North America*. Missouri Botanical Garden, St. Louis, MO.
- Knuth, P. 1908. Handbook of flower pollination. Translated by J.R. Ainsworth Davis. Volume 2. Clarendon Press, Oxford, U.K.
- [KSNPC] Kentucky State Nature Preserves Commission. 1996. Rare and extirpated plants and animals of Kentucky. *Trans. Kentucky Acad. Sci.* 57:69–91.
- Kummer, A. P. 1951. Weed seedlings. Univ. Chicago Press, Chicago, IL.
- Langer, R., and W. Kubelka. 1998. Rhizomanatomie von Vertretern der Gattungen *Potentilla*, *Geum*, *Agrimonia*, and *Filipendula* im Vergleich mit *Radix Tormentillae*. *Sci. Pharm.* 66:387–398. Original not seen; abstract in ArticleFirst, su: Agrimonia; accessed 6 Jun 2000.
- Law, D. 1973. The concise herbal encyclopedia. St. Martin's Press, New York, NY.
- Löve, Á., and D. Löve. 1961. Chromosome numbers of central and northwest European plant species. *Opera Bot.* 5:1–581.
- [LRNP] Lawrence Review of Natural Products. 1995. Agrimony. In Lawrence Review of Natural Products, August 1995.
- Lushchevskaya, G. 1937. Dyestuff-yielding plants in the Ukraine S.S.R. *Chem. Abstr.* 31:6883. (Abstract.)
- Macroberts, M. H., and B. R. Macroberts. 1997. The ecology of *Agrimonia incisa* Torrey & A. Gray (Rosaceae) in the West Gulf Coastal Plain. *Phytologia* 82:114–128.
- Masters, M. T. 1869. Vegetable teratology. Robert Hardwicke, London, U.K.
- McFarland, F. T. 1942. A catalogue of the vascular plants of Kentucky. *Castanea* 7:77–108.
- Medley, M. E. 1993. An annotated catalog of the known or reported vascular flora of Kentucky. Ph.D. dissertation. Univ. Louisville, Louisville, KY.
- Miller, A. B. 1976. Shaker herbs. Clarkson N. Potter, New York, NY.
- Mitchell, J., and A. Rook. 1979. Botanical dermatology. Greengrass, Vancouver, Canada.
- Miyamoto, K., N. Kishi, and R. Koshiura. 1987. Antitumor effect of agrimoniin, a tannin of *Agrimonia pilosa* Ledeb., on transplantable rodent tumors. *Jap. J. Pharmacol.* 43:187–195. [Original not seen; abstract in Medline (su: agrimonia) 87199472. Accessed 4 Jun 2000.]
- M'Murtrie, H. 1819. Sketches of Louisville and its environs. S. Penn Jr., Louisville, KY. ["Florula Louisvillensis," 207–229.]
- Moerman, D. E. 1998. Native American ethnobotany. Timber Press, Portland, OR.
- Moquin-Tandon, A. 1842. Pflanzen-teratologie. Translated from the French by J. C. Schauer. Haude und Spener'sche Buchhandlung, Berlin, Germany.
- Mueller, H. 1883. The fertilisation of flowers. Translated and edited by D. W. Thompson. Macmillan and Company, London, U.K.
- Muenschner, W. C. 1955. Weeds. Cornell Univ. Press, Ithaca, NY.
- Murata, G., and K. Umemoto. 1983. Critical notes on the Asiatic *Agrimonia* with special reference to the distribution of calcium oxalate crystals. *Acta Phytotax. Geobot.* 34:166–177. (In Japanese.)
- Nelmes, E. 1929. *Agrimonia eupatoria* Linn. with white flowers. *J. Bot.* 67:341.
- Nigg, H. N., and D. Seigler. 1992. Phytochemical resources for medicine and agriculture. Plenum Press, New York, NY.
- O'Donovan, W. J. 1942. Dermatitis bullosa striata praten-sis. Agrimony dermatitis. *Brit. J. Dermatol. Syph.* 54: 39–46.
- Ody, P. 1993. The complete medicinal herbal. Dorling Kindersley, London, U.K.
- Phelouzat, R. 1963. Morphologie, Phyllotaxie, ontogénie de trois rosacées. *Agrimonia eupatoria* L., *Poterium sanguisorba* L., *Geum urbanum* L. *Rev. Cytol. Biol. Vég.* 26:101–347, 15 plates. [*A. eupatoria*, 115–293.]
- Pratt, A. 1905. The flowering plants. Grasses, sedges and ferns of Great Britain. Frederick Warne & Company, London, U.K.

- Price, S. F. 1893. Flora of Warren County, Kentucky. Published by the author.
- Quattrocchi, U. 2000. CRC world dictionary of plant names. CRC Press, Boca Raton, FL.
- Radford, A. E., H. E. Ahles, and C. R. Bell. 1968. Manual of the vascular flora of the Carolinas. Univ. North Carolina Press, Chapel Hill, NC.
- Rafinesque, C. S. 1828. Medical flora. Volume 1, A–H. Atkinson and Alexander, Philadelphia, PA.
- [RDA] Reader's Digest Association. 1981. Field guide to the wild flowers of Britain. Reader's Digest Association, London, U.K.
- Reitsma, T. 1966. Pollen morphology of some European Rosaceae. *Acta Bot. Neerl.* 15:290–307.
- Ridley, H. N. 1930. The dispersal of plants throughout the world. L. Reeve and Company, Ashford, Kent, U.K.
- Robertson, C. 1928. Flowers and insects. n.p., Carlinville, IL.
- Robertson, K. R. 1974. The genera of Rosaceae in the southeastern United States. *J. Arnold. Arbor.* 55:303–332, 344–401, 611–662.
- Robertson, S. M. 1973. Dyes from plants. Van Nostrand Reinhold, New York, NY.
- Robinson, B. L. 1900. The nomenclature of the New England agrimonies. *Rhodora* 2:235–238.
- Robinson, B. L. 1901. Further notes on the agrimonias. *Bull. Torrey Bot. Club* 28:295–300.
- Robinson, B. L., and M. L. Fernald. 1908. Gray's new manual of botany. American Book Company, New York, NY.
- Ross-Craig, S. 1956. *Agrimonia eupatoria* L. Drawings of British plants IX Rosaceae (2): plate 9. G. Bell, London, U.K.
- Rydberg, P. A. 1913. *North Am. Fl.* 22(5):391–396.
- Schauenberg, P., and F. Paris. 1977. Guide to medicinal plants. Keats Publishing, New Canaan, CT.
- Schimper, C. 1854. Discentration (unzureichend Fasciation genannt). *Flora* 37:76.
- Schoepf, J. D. 1787. *Materia medica americana*. Reprint, with biography of Johann David Schoepf by E. Krehmers, as *Bull. Lloyd Libr.* 6 (Reproduction Ser. 3). 1903.
- Short, C. W., R. Peter, and H. A. Griswold. 1833. A catalogue of the native phaenogamous plants and ferns of Kentucky. Reprint in R. L. Stuckey, Scientific publications of Charles Wilkins Short. Arno Press, New York, NY, 1978.
- Short, C. W., and R. Peter. 1834. A supplementary catalogue of the plants of Kentucky. *Transylvania J. Med. Assoc. Sci.* 7:598–600.
- Skalický, V. 1973. Amerikanischer Arten der Gattung *Agrimonia* L. ser. Tuberosae ser. nova. *Folia Geobot. Phytotax.* 8:95–104.
- Smith, P. 1812. The Indian doctor's dispensatory. Reprint, with biography of Peter Smith by J. U. Lloyd, as *Bull. Lloyd Libr.* 2 (Reproduction Ser. 2). 1901.
- Svenson, H. K. 1941. Report of the local flora committee. *Torreya* 41:3–7.
- Swanston-Flatt, S. K., C. Day, C. J. Bailey, and P. R. Flatt. 1990. Traditional plant treatments for diabetes. *Diabetologia* 33:462–464. [Original not seen; abstract in Medline (su: Agrimonia, 91006942); accessed 4 Jun 2000.]
- Swink, F., and G. Wilhelm. 1994. Plants of the Chicago region. 4th ed. Indiana Academy of Science, Indianapolis, IN.
- Torrey, J., and A. Gray. 1838–1840. *Agrimonia*. Pages 430–431 in *A flora of North America*. Volume 1. Wiley and Putnam, New York, NY.
- Usher, G. 1974. A dictionary of plants used by man. Hafner Press, New York, NY.
- Voss, E. G. 1985. Michigan flora. Part II Dicots (Saururaceae–Cornaceae). Cranbrook Institute of Science (Bulletin 59) and University of Michigan Herbarium, Ann Arbor, MI.
- Weatherby, C. A. 1924. Two more wool-waste plants from Westford, Mass. *Rhodora* 26:38–39.
- Wood, M. 1997. The book of herbal wisdom. North Atlantic Books, Berkeley, CA.
- Zardini, E. M. 1971. Especies nuevas o criticas de la flora Jujeña II. *Bol. Soc. Argentina Bot.* 14:107–110. [*A. parviflora*.]

NOTE

Barn Owl (*Tyto alba*) Feeding Habits at Yellowbank Wildlife Management Area, Breckinridge County, Kentucky.—This study used pellet analysis to determine feeding habits of barn owls (*Tyto alba*) at the Yellowbank Wildlife Management Area (YWMA), Breckinridge County, Kentucky. We found that barn owl diets at YWMA consisted mainly of voles (*Microtus* spp.). These results are consistent with many past barn owl feeding habit studies.

Past studies suggest that barn owls consume a prevalence of small mammals (1, 2). However, larger animals (e.g., birds, reptiles, and amphibians), and insects (e.g., grasshoppers, beetles) are also often eaten (2, 3). Little is known, however, about barn owl feeding habits in the east-central U.S. We are unaware of any reports of barn owl feeding habits for Kentucky and Indiana, and there are only single reports for southern Illinois (4) and Ohio (5). The purpose of this study was to determine whether prey selection of barn owls in YWMA is consistent with other barn owl feeding habit studies.

Feeding habits of barn owls are generally examined through the dissection of regurgitated pellets. Barn owls consume their prey whole or largely so. Relatively weak digestive fluids secreted in the stomach dissolve the nutritious soft parts of the prey (1, 2). This is followed by the regurgitation of a tightly packed pellet consisting of indigestible fur, bones, insect exoskeleton, and/or feathers. Typically, two pellets are formed and cast each day, ca. 6.5 hr after ingestion of prey. One pellet is cast at a habitual roosting or nesting area, the other at night while the owl is foraging (6). Consumption of more than one meal during a 6.5-hr period may result in the casting of larger pellets containing remains of multiple meals (6). Thus, a record of past meals is created from undigested portions of prey (2, 7). The proportion of prey items captured by barn owls closely matches the proportion regurgitated, providing a relatively unbiased sampling of feeding habits (1, 2).

Pellets were gathered from beneath a nesting platform at YWMA during the 1998 nesting season. Two adult barn owls and seven young regurgitated the pellets.

Each dry pellet was weighed to the nearest 0.1 g, measured along the longest and shortest axes to the nearest 1 mm, soaked in water, then dissected using tweezers and dissecting probes. Skull and lower mandible remnants were counted and identified.

Small mammal capture records for YWMA and a list of mammal species occurring in Breckinridge County (both provided by the Kentucky Department of Fish and Wildlife) were used as a guide for preliminary identification. We used a reference skull collection (Mammal Collection, Southern Illinois University, Carbondale) to assist in species identification of bone fragments (8, 9, 10, 11, 12, 13).

We determined the percentage of each species within pellets by number and by mass, with estimates of mean mass (14, 15). Skull and mandible counts from large pel-

lets (≥ 2.0 g) were compared to those of small pellets (< 2.0 g) with one-tailed *t*-tests adjusted for unequal sample size. Correlation analysis was used to assess the relationship between pellet mass and the number of prey items per pellet. Species proportions between small and large pellets were examined with chi-square, with a 2.0 g cut-off arbitrarily chosen after inspection of the distribution of pellet masses.

We examined 52 large and 151 small pellets for a total of 203 pellets. An average dry pellet weighed 1.6 ± 0.1 g, and was 29.8 ± 0.5 mm long \times 20.9 ± 0.4 mm wide. Small pellets contained fewer skulls (mean = 0.63) than large pellets (mean = 1.49); $t_{67} = -6.79$, $P < 0.001$. Small pellets also had fewer mandibles (mean = 0.94) than large pellets (mean = 3.06; $t_{66} = -7.80$, $P < 0.001$).

The most common genus among all pellets was *Microtus* (64%), primarily *M. ochrogaster* ($n = 87$), and *M. pinetorum* ($n = 36$; Table 1). Southern bog lemmings (*Synaptomys cooperi*; $n = 15$) were also prevalent. Species diversity increased as a function of pellet mass ($r^2 = 0.24$, $P < 0.001$). We were unable to classify remains from 29 of the 203 (14%) pellets. The proportion of unidentified specimens in small pellets was higher than in large pellets (27.9 vs. 3.8%, respectively; $\chi^2 = 20.02$, $P < 0.001$). The proportion of unidentified microtines in small pellets was nearly double that of large pellets (13% vs. 7.5%), though this difference was not significant, ($\chi^2 = 1.60$, $P < 0.21$). Composition of identified species was similar between small and large pellets, aside from a greater number of southern bog lemmings in the smaller pellets (13.0 vs. 2.0; $\chi^2 = 6.35$, $P < 0.01$). Pellets were rarely devoid of bones ($n = 4$). The only apparent difference in prey composition between pellet class sizes was in proportion of southern bog lemmings.

Barn owls are opportunistic foragers. Prevalence of prairie and woodland voles in the barn owl diet is likely a function of their relative ease of capture, concurrence of predator and prey activity periods, and prey abundance. Barn owls are most active at night and favor open country for foraging (3). Likewise, prairie and woodland voles remain on the ground (versus escaping up into the trees) when pursued. Woodland voles are also common in most stages of forest succession (12). Our data suggest that populations of shrews (*Blarina* sp.; *Sorex* spp.) and harvest mice (*Reithrodontomys* spp.) are not active during the same time period that barn owls hunt, do not forage in the same areas as barn owls, or use specific habitat components successfully to evade predation. In conclusion, our results indicate a preponderance of microtines in the diet of barn owls at YWMA and are consistent with other barn owl feeding habit studies (2, 4, 5, 16, 17).

We thank Dr. G. A. Feldhamer, and J. C. Whittaker, Department of Zoology, SIUC, and Dr. Alan Woolf, Co-operative Wildlife Research Laboratory and Department

Table 1. Numbers and percentages of known prey species found in 203 pellets from a pair of barn owls and their seven young at Yellowbank Wildlife Management Area, Breckinridge County, Kentucky.

Species (mean live mass [g])	Common name	Number	% by number	% by mass ¹
<i>Microtus ochrogaster</i> (42.5)	Prairie vole	87	50	61
<i>Microtus pinetorum</i> (32)	Woodland vole	36	21	19
<i>Synaptomys cooperi</i> (35.5)	Southern bog lemming	15	9	9
<i>Blarina brevicauda</i> (21.5)	Short-tailed shrew	9	5	3
<i>Mus musculus</i> (20.5)	House mouse	8	5	3
<i>Cryptotis parva</i> (5.3)	Least shrew	7	4	<1
<i>Microtus pennsylvanicus</i> (45)	Meadow vole	7	4	5
<i>Reithrodontomys megalotis</i> (13)	Harvest mouse	1	<1	<1
Unidentified bird ²		4	2	NA
Total ³		174		

¹ For identified specimens only.
² One specimen was likely a red-winged blackbird (*Ageaius phoeniceus*).
³ Excluding plant and insect contents.

of Zoology, SIUC, and numerous volunteers for laboratory assistance.

LITERATURE CITED. (1) Bunn, D. S., A. B. Warburton, and R. D. S. Wilson. 1982. The barn owl. Buteo, Vermillion, SD. (2) Taylor, I. 1994. Barn Owls, predator-prey relationships and conservation. Cambridge University Press, Cambridge, UK. (3) Bent, A. C. 1938. Life histories of North American birds of prey. Bull. U.S. Natl. Mus. 170:140–153. (4) Feldhamer, G. A. 1985. Summer feeding habits of barn owls (*Tyto alba*) from White County, Illinois. Trans. Illinois Acad. Sci. 78:133–137. (5) Colvin, B. A., and E. B. McClean. 1986. Food habits and prey specificity of the common barn owl in Ohio. Ohio J. Sci. 86:76–80. (6) Marti, C. D. 1992. Barn owl. Pages 1–16 in A. Poole, P. Stettenheim, and F. Gill (eds). The birds of North America. The American Ornithologists' Union and Acad. Nat. Sci. Philadelphia, Philadelphia, PA. (7) Sparks, J., and T. Soper. 1970. Barn owls, their natural and unnatural history. Taplinger, New York. (8) Barbour, R. W., and W. H. Davis. 1974. Mammals of Kentucky. Univ. Kentucky Press, Lexington, KY. (9) Martin, R. A. 1974. Fossil mammals from the Coleman IIA fauna, Sumter County. Pages 35–99 in S. D. Webb (ed). Pleistocene mammals of Florida. Univ. Florida Press, Gainesville, FL.

(10) Hazard, E. B. 1982. The mammals of Minnesota. Univ. Minnesota Press, Minneapolis, MN. (11) Carraway, L. N. 1995. A key to recent Soricidae of the western United States and Canada based primarily on dentaries. Occas. Papers Mus. Nat. Hist. Univ. Kansas. 175. (12) Hoffmeister, D. F. 1989. Mammals of Illinois. Univ. Illinois Press, Urbana, IL. (13) Feldhamer, G. A., L. C. Drickamer, S. H. Vessey, and J. F. Merritt. 1999. Mammalogy. McGraw-Hill, Boston, MA. (14) Burt, W. H., and R. P. Grossenheider. 1980. A field guide to the mammals, North America north of Mexico. Houghton Mifflin, New York. (15) Whitaker, J. O., Jr. 1996. National Audubon Society field guide to North American mammals. Knopf, New York. (16) Wallace, G. J. 1948. The barn owl in Michigan. Page 61 in Craighead, J. J., and F. C. Craighead, Jr. (eds). Hawks, barn owls, and wildlife. Dover, New York. (17) Craighead, J. J., and F. C. Craighead, Jr. 1969. Hawks, barn owls, and wildlife. Dover, New York.—**Anita T. Morzillo**, Department of Zoology; **Hetti A. Brown**, Department of Biological Sciences; **Wayne E. Thogmartin**, Department of Zoology & Cooperative Wildlife Research Laboratory; and **Jennifer H. Herner-Thogmartin**, Cooperative Wildlife Research Laboratory, Southern Illinois University, Carbondale, Illinois 62901.

Abstracts of Some Papers Presented at the 1999 Meeting of the Kentucky Academy of Science

AGRICULTURAL SCIENCES

Pesticide residue in soil and runoff: measurement and mitigation. GEORGE F. ANTONIOUS, Community Research Service, Atwood Research Facility, Department of Plant and Soil Science, Kentucky State University, Frankfort, KY 40601.

Soil erosion and runoff are some of the major means by which pesticides from agricultural fields enter streams, ponds, or lakes. The Water Quality Project at Kentucky State University (KSU) is evaluating best management practices for the growing of vegetable crops on highly erodible land (10% slope). Studies were conducted to determine the influence of landscape features on pesticide movement into runoff and infiltration water. Soil treatments (black plastic mulch and living fescue mulch) were used to reduce soil erosion and surface water runoff. Pesticides infiltration into the vadose zone were monitored using pressure-vacuum lysimeters ($n = 27$). A tipping bucket metering apparatus was used to collect runoff water following natural rainfall events. The impact of the soil mulches on movement of clomazone (a soil applied herbicide), dacthal (a pre-emergence non-systemic herbicide), and endosulfan (an insecticide) was measured under field conditions. Black plastic covers had no clear effect on reducing runoff volume or concentration of clomazone in runoff, while grass strips of 30-cm wide were very effective at reducing amounts of sediment in runoff. Plots planted with pepper intercropped with tomato as cover crop had 72% less runoff water and 79% less runoff sediment compared to plots planted with pepper only. Results indicated the vertical movement of clomazone, dacthal, and endosulfan through the soil into the vadose zone. Cultivation of turf reduced runoff but did not reduce leaching of pesticides into the vadose zone. Our future objective at KSU is to study the potential of using soil amendments to improve soil quality, detoxify contaminants, and reduce erosion.

Identification of molecular markers that segregate in a simple Mendelian fashion in controlled crosses of pawpaw (*Asimina triloba*). SHAWN BROWN,* TERA M. BONNEY, SNAKE C. JONES, and KIRK W. POMPER, Kentucky State University, Atwood Research Facility, Frankfort, KY 40601-2335.

The pawpaw (*Asimina triloba*) is a native plant found mainly in the southeastern and eastern United States. Its fruit has great potential as a new high-value crop in these regions. Although there are about 45 named pawpaw cultivars, breeding for improvement of desirable traits, such as improved fruit size and quality, is desirable. Our long-term goal is to utilize molecular marker systems to identify markers that can be used for germplasm diversity analysis and for the construction a molecular genetic map, where

markers are correlated with desirable pawpaw traits. The objective of this study was to identify random amplified polymorphic DNA (RAPD) markers that segregate in a simple Mendelian fashion in a controlled *A. triloba* cross. DNA was extracted from young leaves collected from field-planted parents and 20 progeny of the cross 1-7-1 \times 2-54, as well as 10 progeny of the reciprocal cross. The DNA extraction method used gave acceptable yields of about $7 \mu\text{g g}^{-1}$ of leaf tissue. Additionally, sample 260/280 ratios were about 1.4, which indicated that the DNA was of high enough purity to be subjected to the RAPD methodology. Screening of 10-base oligonucleotide RAPD primers with template DNA from the parents and progeny of the cross is proceeding in an effort to identify RAPD markers that segregate in a simple Mendelian fashion.

The effect of transport density on survival of juvenile freshwater prawns (*Macrobrachium rosenbergii*). SHAWN COYLE, JAMES H. TIDWELL, AARON VANARNUM,* and CHARLES WEIBEL, Aquaculture Research Center, Kentucky State University, Frankfort, KY 40601.

In production of the freshwater prawn *Macrobrachium rosenbergii*, prawns (≥ 0.3 g) are nursed in indoor tanks then transported to ponds for growout. Stress during transport can produce immediate and undetected mortality after pond stocking. This study was designed to evaluate the effect of density on transport survival. Nine replicate styrofoam transport containers were prepared. Each contained a plastic bag with oxygen-saturated 22°C water with an atmosphere of 10-liter pure oxygen. Juvenile prawns weighing 0.26 ± 0.02 g were randomly stocked into each of three replicate transport containers at 10, 25 or 50 g/liter of water, then sealed for 8 hours (maximum in-state transport period). At 8 hours post-stocking, bags were opened, water was sampled, and live and dead animals were separated and counted. Total ammonia-nitrogen and nitrite-nitrogen were significantly higher ($P < 0.05$) in containers stocked at 50 g/liter than in containers stocked at either 10 or 25 g/liter, which were also significantly different ($P < 0.05$). Dissolved oxygen was significantly lower ($P < 0.05$) in transport containers stocked at 50 g/liter (1.3 mg/liter) than those stocked at 25 g/liter or 10 g/liter (1.6 mg/liter and 3.2 mg/liter, respectively), which were also significantly different ($P < 0.05$). Survival was significantly reduced ($P < 0.05$) in transport containers stocked at 50 g/liter (86.6%). Survival in containers stocked at 25 g/liter (93%) was significantly lower than containers stocked at 10 g/liter (97.2%). These data indicate that transport densities greater than 10 g/liter should be avoided for transport ≥ 8 hours.

Suitability of the copepod *Orthocyclops modestus* as a live food for larval freshwater prawns, *Macrobrachium ro-*

senbergii. SHAWN COYLE,* JAMES H. TIDWELL, AARON VANARNUM, and CHARLES WEIBEL, Aquaculture Research Center, Kentucky State University, Frankfort, KY 40601.

The cyclopoid copepod *Orthocyclops modestus* was evaluated for its suitability as a live food for larval freshwater prawns (*Macrobrachium rosenbergii*). *Orthocyclops modestus* was identified in preliminary screening as one of the few indigenous zooplanktors which tolerated the temperature (28–30°C) and salinity (10–14 ppt) conditions of larval prawn culture. To evaluate the suitability of these copepods as live food for larval prawns, mixed zooplankton were collected from a reservoir with a 250 µm zooplankton net. Zooplanktors were held at 10 ppt salinity for 24 hours to remove cladocerans and then screened (335 µm) to remove rotifer species leaving only copepods. The test system consisted of nine individual 250 ml rearing units in a recirculating system, with three replicates of each of three treatments. Treatment 1 contained only larval prawns (five 10-d old larvae, Stage 5.4 ± 0.9), Treatment 2 contained only copepods (185), and Treatment 3 a combination (5 larval prawns and 185 copepods). Densities for prawns were based on recommended prawn hatchery practices. Densities for copepods were based on recommended artemia feeding rates for Stage 5 prawn larvae. After 48 hours, prawn survival in Treatment 3 (87%) was significantly lower ($P < 0.05$) than in Treatment 1 (100%). Copepod survival was not significantly different between treatments (93.7%) indicating copepods were not consumed. Reduced prawn survival in Treatment 3 was likely due to high energy demands or physical trauma as prawn larvae attempted to capture active and well-armored copepods. It appears that indigenous zooplankton show little promise as live foods in prawn hatchery production.

Use of hempseed meal, poultry by-product meal, and canola meal in practical diets without fish meal for sunshine bass. ANN M. MORGAN,* CARL D. WEBSTER, KENNETH R. THOMPSON, and EBONY J. GRISBY, Aquaculture Research Center, Kentucky State University, Frankfort, KY 40601.

Sunshine bass (*Morone chrysops* × *M. saxatilis*) is one cross of hybrid striped bass. Diets for sunshine bass use high percentages of fish meal (FM); however, FM is the most expensive ingredient in aquaculture diets. If FM can be replaced, diet costs may decrease. Four practical floating diets were formulated to contain 40% protein, similar energy levels, and without FM. A fifth diet was formulated to contain 30% FM and served as the control diet. Ten fish (21 g) were stocked into each of twenty 110-liter aquaria and were fed twice daily (07.30 and 16.00 hr) for 10 weeks. Diets were formulated to contain: Diet 1, soybean meal (SBM) and meat-and-bone meal (MBM); Diet 2, SBM + MBM + hempseed meal (HSM); Diet 3, SBM and poultry by-product meal (PBM); and Diet 4, and SBM + MBM + canola meal (CM). At the conclusion of the feeding trial, percentage weight gain of fish fed Diet

1 was significantly ($P < 0.05$) higher (299%) compared to fish fed Diet 3 and Diet 4, but not different from fish fed Diet 2 and Diet 5. Percentage survival, amount of diet fed, and hepatosomatic index (HSI) of sunshine bass were not different ($P > 0.05$) among treatments and averaged 95%, 111 g of diet/fish, and 2.0% of body weight, respectively. Results from the present study indicate that diets without FM have potential for use in growing juvenile sunshine bass. Further research needs to be conducted on the diet formulations used in the present study and should be conducted in ponds.

The Kentucky State University pawpaw (*Asimina triloba*) project. KIRK W. POMPER,* SNAKE C. JONES, EDDIE B. REED, and TERA M. BONNEY, Kentucky State University, Atwood Research Facility, Frankfort, KY 40601-2355.

Kentucky State University (KSU) has had a comprehensive pawpaw (*Asimina triloba*) research project since 1990, with the goal of developing the pawpaw into a new high-value tree fruit crop for limited resource farmers in Kentucky. An overview of recent developments at KSU concerning pawpaw variety trials, propagation, web site development, and germplasm collection and assessment, was presented. A pawpaw regional variety trial (RVT) was planted at KSU in 1998, consisting of 8 replicate trees of 28 standard pawpaw cultivars and advanced selections from the PawPaw Foundation breeding program. The RVT establishment rate, flowering, and growth data by variety was reported. The positive influence of shade on the growth and development of pawpaw seedlings in container production was discussed. A web site, <http://www.pawpaw.kysu.edu>, has been developed and expanded for the dissemination of information on pawpaw to scientists, commercial growers and marketers. The National Clonal Germplasm Repository for *Asimina* spp. is located at KSU and hence, germplasm evaluation, preservation, and dissemination are a high priority for our program. In the spring of 1999, volunteers collected pawpaw leaf samples from 270 trees in 17 different states that will be used in molecular marker methodologies in order to assess genetic diversity across the pawpaw's native range. Pawpaw seedlings with promising fruit characteristics have been identified in our germplasm collection and have been propagated for further evaluation as potential cultivars for release by KSU.

Relative effectiveness of plant and animal source oils for control of air breathing insects. LEIGH ANNE VITATOE,* AARON VANARNUM, SHAWN COYLE, and JAMES TIDWELL, Aquaculture Research Center, Kentucky State University, Frankfort, KY 40601.

Freshwater prawn juveniles (*Macrobrachium rosenbergii*) are stocked into ponds at extremely small sizes (0.2–0.5 g) and predation by air breathing insects can be a significant problem. The use of petroleum products to create a thin surface film and prevent insect respiration is an effective control but causes environmental concern. If

proven effective, plant- or animal-based oils may be safer. Menhaden fish oil (MO) and corn oil (CO) were evaluated at two application rates and compared to previously recommended petroleum product mixes for their ability to eliminate air-breathing insects. Petroleum product applications (controls) included 2:1 motor oil/diesel fuel combination (PCI) and 1:20 motor oil/diesel fuel combination (PCII) (previously recommended procedures). Glass aquaria with 0.107 m² surface area were used and filled with 6 liters of reservoir water. Each tank was stocked with five adult notonectids. Low rate applications were applied at 0.01 ml/m² and high rate applications at 0.03 ml/m². There were three replications per treatment. Control tanks were stocked but not treated with oil. At the low rate the PCII and MO treatments both produced complete mortality by 2 hours post treatment while treatments PCI and CO did not result in complete mortality. At the high rate there was no significant difference ($P > 0.05$) among treatments in amount of time required to attain complete mortality. Menhaden fish oil appears to be an effective alternative to petroleum products for control of predaceous air breathing insects in larval shrimp and fish ponds. At a high rate of application corn oil may also be effective.

The effect of water temperature on the survival of adult freshwater prawns (*Macrobrachium rosenbergii*) held in tanks. CHARLES WEIBEL,* JAMES H. TIDWELL, SHAWN COYLE, and AARON VANARNUM, Aquaculture Research Center, Kentucky State University, Frankfort, KY 40601.

Pond production of freshwater prawns (*Macrobrachium rosenbergii*) is becoming increasingly popular in Kentucky. As a seasonal crop, prawns must be harvested by mid-October to prevent losses. However, the highest demand for the product is during the holiday period of November through December. The ability to hold live freshwater prawns would allow producers to address this lucrative market. Temperature directly affects the metabolism of poikilothermic animals and reduced temperatures might increase survival under stressful conditions. Also, females carrying eggs at harvest are not considered desirable by some consumers and temperature could affect this trait. To address this, the effect of temperature on survival of adult freshwater prawns was evaluated under controlled conditions in tanks for 11 weeks. Freshwater prawns recently harvested from ponds (21.3 ± 1.7 g) were randomly stocked into nine 5700-liter tanks at 500 prawns/tank. There were three replicate tanks per temperature (17, 20, and 23°C). Prawns were fed a percentage of body weight at maintenance levels. After 76 days, average weight was not significantly different ($P > 0.05$) between treatments. Survival was significantly higher ($P < 0.05$) for animals held at 20° and 23°C (58 and 56%, respectively) than at 17°C (29%). The percentage of berried (egg carrying) females was significantly greater ($P < 0.05$) at 23° (13.2%) than at 20° or 17°C (0.2 and 0%, respectively). These data indicate that 20°C may be optimum for holding freshwater

prawns for market if berried females are undesirable. Holding temperatures near 17°C appear to represent stress conditions and result in high mortality.

BOTANY & MICROBIOLOGY

Inheritance of morphological and physiological characteristics in *Taraxacum officinale*. ANTON M. CLEMMONS* and DAVID L. ROBINSON, Department of Biology, Bellarmine College, Louisville, KY 40205.

Dandelion (*Taraxacum officinale*), an asexual species, produces achenes (seeds) apomictically. Three experiments were performed to explore the genetic variability and heritability of various morphological/ physiological traits that occur in natural populations. In the first study, achenes from a single dandelion population were subjected to a warm temperature treatment (37°C) for different time periods (0, 3, 4, 6, 8, 10, 14, 16 d), followed by a 4-d incubation at 21°C, and a final cold treatment (5°C). Germination was assessed at the end of each temperature treatment. The experimental control (constant 21°C) exhibited the highest percent germination. In the other treatments, a positive correlation was found between the percent of ungerminated achenes and the duration of exposure to high temperature. To examine the heritability of achene heat tolerance, germinated achenes exposed to 8 d at 37°C, and then 21°C, were collected, grown to maturity, and allowed to flower and set seed. Achenes from these heat-tolerant plants (as well as control plants) were grown to maturity to evaluate the next generation. In the second study, achenes collected from dandelion populations occurring in five U.S. states were germinated and grown to maturity in a controlled environment. Analysis of leaf morphology revealed more variability between the five populations than within them. In the third experiment, achenes from 19 different fasciated (deformed) plants were germinated and grown in a controlled environment to examine the inheritance of their expressed fasciation. These experiments help to delineate the amount of genetic diversity in dandelion populations.

Biological effects of volatile emissions from cut turf. MARIA L. DAVIS* and DAVID L. ROBINSON, Biology Department, Bellarmine College, Louisville, KY 40205.

Current research on the volatile emissions emanating from recently-mowed turfgrass indicates that these gases may have a significant impact on the environment. The primary purpose of this study was to observe the effect of gaseous emissions from different species on the rate of seed germination. In the first study, dandelion (*Taraxacum officinale*) and white snakeroot (*Ageratina altissima* [*Eupatorium rugosum*]) seeds were placed into sealable, plastic bags containing the freshly harvested foliage (cut into ca. 4 cm lengths) from a species of plant that commonly grows in turf. Seeds were treated with the emissions from one of seven plant species: *Cynodon dactylon* (L.) Pers., *Lolium perenne*, *Leptochloa fascicularis*, *Taraxacum officinale*, *Trifolium repens*, *Glechoma hederacea*, or *Plantago lanceolata*. Germination rates were examined over a 18-d

period, and the foliage-gas treatments that incurred the greatest effects were examined in more detail in a second experiment. In that study, *A. altissima* seeds were germinated in a replicated trial in the presence of cut foliage from one of four different species (*L. perenne*, *T. officinale*, *T. repens*, *P. lanceolata*) or an experimental control. No statistically significant differences were observed for rate of germination in any of the foliage treatments (including the control). In addition, the effect of these gases on crickets, a common grass-dwelling insect, will be discussed.

Identification of fecal coliform species from Lee's Branch at Midway, Kentucky. BEVERLY W. JUETT,* DEBORAH EVEN, and GLENDA MARKER, Department of Biological Sciences, Midway College, Midway, KY 40347.

Coliform species were isolated and identified from 60 total coliform counts performed in Lee's Branch at Midway, KY. Water sampling was conducted six different times beginning in July 1996 and ending in September 1997. Total coliform counts were determined by the Standard Total Coliform Membrane Filter Procedure. Coliform species isolated from Endo agar plates were transferred to trypticase soy and MacConkey agar. The isolates were identified by conventional biochemical methods and Enterotube II. Identification of bacterial species in stream water provides basic knowledge of the microbial environment of freshwater and may serve as baseline data in determining if the fecal contamination of this stream is from animals or humans.

Continuation of a study on inheritance of achene characteristics in *Ageratina altissima*. JOANN M. LAU* and DAVID L. ROBINSON, Department of Biology, Bellarmine College, Louisville, KY 40205.

Seed dormancy is a powerful means by which plants control when and where they occur. Three major sources for a population's variability for dormancy are genetic diversity, somatic polymorphism, and microsite/temporal/biological variability. The goal of this research was to explore the relative importance of these sources in regulating achene (seed) germination and dormancy in white snakeroot (*Ageratina altissima* [*Eupatorium rugosum*]). Last year, white snakeroot achenes were selected for different germination and dormancy traits, grown to maturity and allowed to reproduce. The progeny (achenes) of these selections were then examined for different germination characteristics. Most (54%) of the progeny germinated before any cold treatment, whereas 12% germinated after a single cold treatment. Although, on average, there were no noticeable differences between the parental groups, there were obvious differences between individual selections. For instance, one plant (from a non-cold-requiring achene) produced achenes that exhibited 100% germination at room temperature, whereas another plant's progeny (selected for germination after 1 cold treatment) exhibited 32, 20 and 16% germination after 0, 1, and 2 cold

treatments, respectively. In another study, the effect of achene size on germinability was examined by partitioning achenes from a single population into four size categories. Even though the weight of the largest achenes was more than double the smallest there were no statistically significant differences in cumulative germination between any of the size categories. Therefore, if there is somatic polymorphism for achene dormancy in white snakeroot it may involve characteristics other than achene size.

CELLULAR & MOLECULAR BIOLOGY

The GABAc rho1 and rho2 subunit genes are differentially expressed during pre- and postnatal development in the mouse. CHRISTY COLE,* Transylvania University, Lexington, KY 40508; and MAUREEN McCALL, University of Louisville, Louisville, KY.

Most inhibitory neurotransmission in the central nervous system (CNS) is mediated either by glycine or gamma aminobutyric acid (GABA). GABA inhibition is mediated by one of three receptors: GABAA, GABAB, or GABAc. In the rodent, the GABAc receptor regulates the flow of chloride ions in the bipolar cells of the retina. The GABAc receptor may be made up of three types of subunits: rho1, rho2, and rho3. The GABAc rho2 subunit is predominant throughout the CNS, however, the GABAc rho1 subunit seems to be predominant in only the bipolar cells of the retina. These subunits are expressed at different times in the development of the mouse. To determine at which point in the development of the mouse the GABAc rho1 and rho2 subunits are expressed, total RNA was extracted from mice at embryonic ages (E): E12.5, E14.5, E17.5 and from retinas of mice at postnatal ages (P): P1, P6, P13, P20. cDNA was reverse transcribed from all RNA samples and used in a PCR based strategy to determine the time course of the expression of both the GABAc rho1 and rho2 subunit genes. As a control for the amount of cDNA transcribed from each RNA sample, beta-actin was amplified from each cDNA sample. The expression of all three genes were tested individually using PCR, and the amplified products were visualized using electrophoresis. We have found that the GABAc rho1 gene and the rho2 gene are differentially expressed during pre- and postnatal development of the mouse. The GABAc rho1 gene appears to begin to be expressed between P1 and P6. The GABAc rho2 expression is present in the youngest sample that we tested, E12.5. Although the GABAc rho1 and rho2 genes are thought to assemble into functional receptors in the adult, their differential expression during development suggests that the GABAc rho2 gene may play a different role in the developing embryo and may assemble homomeric receptors or associate with other GABA subunits to assemble functional receptors. These results will also aid in further research with transgenic and knockout mice which are being tested to determine the function of the GABAc receptor in retinal processing.

CHEMISTRY

Photoacoustic measurements in biological tissues and fluids. ANGELA L. NEWCOMB,* Department of Science, Campbellsville University, Campbellsville, KY 42718; and JOEL MOBLEY and TUAN VO-DINH, Life Sciences Division, Oak Ridge National Laboratory, Oak Ridge, TN 37831.

The photoacoustic (PA) effect is the generation of acoustic waves by electromagnetic radiation. The magnitude of the PA response in a material is determined by its optical absorption. We have investigated the thermally-mediated PA response of mammalian brain tissue and glucose solutions to explore the utility of PA methods for biomedical sensing applications. We measured the PA spectra of tissues from the cerebrum of sheep between 500–700 nm using a tunable pulsed laser for excitation. We were able to differentiate between the gray and white matter in our samples, which could be useful in minimally invasive surgery. We were also able to detect the presence of glucose in solution, which may lead to a noninvasive method of measuring blood sugar.

HEALTH SCIENCES

Analysis of a functional digital model of the mammalian basilar membrane. SHELLY FERRELL,* MATTHEW E. KOGER, DAVID RICE, KENNETH M. MOORMAN, and PEGGY SHADDUCK PALOMBI, Transylvania University, Lexington, KY 40508.

The human auditory system functions as a processor of sound. The processing occurs in an analog setting where the sound stimulus is generated from a source and is directed toward the ear. From the pinna, the sound travels in the outer ear to the middle ear, through the cochlea where the basilar membrane vibrates to stimulate the hair cells. In this pathway the signal is transduced from a wave front to an electrical signal. This is also the critical point where the information is translated into a signal that the brain can process. The sound signal has a non-linear relation to the cochlear output. For a fully functioning computer model to simulate the process of basilar membrane function, it must contain the non-linear properties that the basilar membrane incorporates into the signal processing. Other minor inputs to the main sound wave are from sources such as pinna vibration, skull vibration, ossicular vibration, and residual tympanic membrane vibration. We attempted to evaluate a “best” computer model of auditory processing for accurate models that incorporate consistent non-linear properties and accurately use the other input conditions to fine tune and purify the simulation. We determined that the LUTEar model developed by Ray Meddis and his colleagues, contains a developed program that has moderately accurate non-linear properties. The consistency of the digital diagrams compared to physiological data is not as accurate as we would like. The overall processing shape is similar; however, the actual numbers are not within an acceptable percent error. Our next steps will be to reprogram portions of the models to achieve a

more accurate model, to correct minor non-linear property errors, and to increase incorporation of other stimuli to the processing output.

Examination of the validity of the pre-emphasis filter in an auditory system model. MATTHEW E. KOGER,* SHELLY FERRELL, DAVID RICE, KENNETH M. MOORMAN, and PEGGY SHADDUCK PALOMBI, Transylvania University, Lexington, KY 40508.

LUTEar, a mammalian auditory system computer model, was examined to determine whether it could be manipulated to correctly model aged hearing. The validity of the pre-emphasis filter in LUTEar was investigated using previously published physiological data. The pre-emphasis filter corresponded with the outer and middle ear in the mammalian auditory system. It takes the input signal and passes it through a mathematical filter. This output is next sent to the basilar membrane filter. An investigation of the literature led to an understanding of the structure and function of the outer and middle ear; based on this understanding, physiological data was gathered from the literature and compared to data from LUTEar gathered by Meddis and Hewitt (1991). It was determined that LUTEar amplified incoming sound in the outer and middle ear ten to twenty decibels less than what physiological data has found. However, it was determined that the basic filter shape of the amplification curve of the pre-emphasis filter closely matched the curve in physiological data. Next, the feasibility of altering the parameters of the pre-emphasis filter for aged hearing was explored. The pre-emphasis filter is a mathematical equation derived from a digital band-pass filter. The difficulty in altering this equation to accurately represent aged hearing was centered in the non-physiological based variables in the filter. It was determined that a greater study of this was needed. Therefore, changes that incorporate biological variables will have to be made to the digital band-pass filter in order to correctly model aged hearing.

A radioprotective drug combination in mice. K. S. KUMAR, V. SRINIVASAN, D. L. PALAZZOLO,* E. P. CLARK, and T. M. SEED, Radiation Medicine Department, Armed Forces Radiobiology Research Institute, Bethesda, MD 20889; and Pikeville College School of Osteopathic Medicine, Pikeville, KY 41501.

Thiol drugs are very effective radioprotective agents. However, the doses required to protect are also very toxic. One approach to minimize toxicity is to combine low doses of thiols with other non-thiol radioprotective agents. The thiol drug used was S-2-(3-methylaminopropylamino)-ethylphosphorothioic acid (WR-3689; 50 mg/kg), a methylated derivative of amifostine, which is used in chemotherapy to reduce the cis-platinum-induced nephrotoxicity. The non-thiols used were monophosphoryl lipid A (MPL, 0.5 mg/kg), an immunomodulator, and two prostaglandins—iloprost (ILO, 0.1 mg/kg), and misoprostol (MIS, 0.1 mg/kg). Individually, these doses of WR-3689 and MPL are known to be non-toxic, while ILO and

MISO are known to be toxic. In this study, male CD2F1 mice were given these agents intraperitoneally (IP) 30 minutes before irradiation with 10 Gy of ^{60}Co at 1 Gy/minute, and survival was monitored for 30 d. When given individually, none of drugs were protective, as indicated by 0% survival. However, when all four agents were combined and given as a single IP injection, the survival rate increased to 50% and a dose reduction factor of 1.23 was calculated indicating significant radioprotection. In addition, the combined drug treatment appeared to be less toxic. These results indicate that the toxicity of radioprotectants at higher doses can be reduced by combining them at lower doses without compromising the radioprotective efficacy.

MATHEMATICS

Superficial coset curiosities. JAMES B. BARKSDALE JR., Department of Mathematics, Western Kentucky University, Bowling Green, KY 42101.

This presentation exhibits several rudimentary propositions concerning coset notions from elementary group theory. Superficial curiosities which result from slight statement modifications of these fundamental theorems and relationships are then noted and discussed. Such presentation content could serve as an enrichment theme (or as a special project topic) for undergraduate mathematics courses.

Open mindedness to low-tech teaching methods. AUSTIN FRENCH, Department of Mathematics/Physics/CSC, Georgetown College, Georgetown, KY 40324.

An effective teaching method involving (1) no note-taking by students (but the students have a clear set of notes), (2) no homework grading by the instructor for outside work, (3) a text that costs a maximum of \$9, (4) a system where it is extremely hard for a student to cheat and where the student's grade measures what the student knows (not that the student was in a group with someone that knew something and they got that student's grade), (5) a mixture of practice and creativity is expected by the student, (6) knowing absolutely perfectly some hard problem types is rewarded, (7) where two overhead projectors are used, and (8) transparencies are made from neat pencil-written notes . . . will be introduced. The concept of overheads as the text will be shown. Surgical strike class questions, microquizzes, presentations, and jugular problems will be described in this unique grading system, which is simply fun to teach by and does not squeeze all of the blood out of the teacher's turnip to use this method. Two overheads are used in conjunction with transparencies from pencil-written overheads comprising the text for the course. This talk should help you to inform the technologically gullible that just because something involves high-tech methods, it does not necessarily mean it is best to use technology and that all need to be open-minded about other teaching methods.

PHYSICS & ASTRONOMY

Searching for hydrogen gas in shell galaxies. SEPPO LAINE,* Department of Physics and Astronomy, University of Kentucky, Lexington, KY 40506; STEPHEN T. GOTTESMAN and KARL E. HAISCH JR., Department of Astronomy, University of Florida, Gainesville, FL 32611; and BENJAMIN K. MALPHRUS, Department of Physical Sciences, Morehead State University, Morehead, KY 40351.

Recent images of the hydrogen line emission from shell galaxies have given new clues about the origin of the shells. Detection of gaseous material associated with these optical features gives us an opportunity to investigate the formation mechanism of shells and even of the associated lenticular or elliptical galaxies. Therefore, we sought to expand the sample of shell galaxies that could be mapped in the 21-cm hydrogen line. We used the 140-ft telescope of the National Radio Astronomy Observatory at Green Bank, WV to attempt to detect the 21-cm emission line in 9 shell galaxies. The observations were centered at the frequency of the spectral line, corresponding to the systemic velocity of the underlying galaxy. The total on-source integration times varied between 3 and 11 hours. We detected emission from the direction of two shell systems, NGC 3610 and NGC 4382. However, NGC 4382 has a nearby disk galaxy within the 20 arcmin beam of the telescope. Therefore, the more likely candidate for detection of gas associated with shells is in NGC 3610. Since the signal extends over a rather narrow range of velocities and the optical images show a small, possibly interacting galaxy within the radio telescope beam, the explanation for this detection is unclear. Recent Very Large Array 21-cm observations of NGC 3610 should resolve the source of the hydrogen line emission.

PHYSIOLOGY & BIOCHEMISTRY

Effect of Tamoxifen, Genistein, and vitamin E on the activity of the cysteine proteases Cathepsin L and Cathepsin B and their endogenous inhibitors in human androgen-independent prostate cancer cell lines. T. BURCHIO,* E. HUGO, M. MARKEY, B. PHILLIPS, A. BURNS, D. DARIA, J. THOMPSON, T. HOLDEN, E. McDONOUGH, G. HENSON, D. FRITZ, B. HURST, and J. H. CARTER, Wood Hudson Cancer Research Laboratory, Newport, KY 41071.

Metastatic prostate cancer is a leading cause of death in men. Initially, most forms of this cancer are repressed by the removal of androgens and several treatment courses are based on this phenomenon. Growth inhibition by androgen removal, however, is often transient, with the carcinoma growth becoming independent of exogenous androgens. The proliferation of tumor cells correlates with the levels of the lysosomal proteases Cathepsin B (CB) and Cathepsin L (CL) as well as the endogenous inhibitors (CPI) of these enzymes. The role of these proteins in the progression of prostate carcinoma (CaP) is most likely one of protein turnover, however, we cannot rule out the pos-

sibility of some interaction with the process of metastasis. We have previously demonstrated that Tamoxifen ((Z)-2-[4-(1,2-diphenyl-1-butenyl) phenoxy]-N, N-dimethyl-ethanamine 2-hydroxy-1,2,3- propanetricarboxylate) (Tam), an antiestrogen, Genistein (4',5,7-trihydroxyisoflavone) (Gen), a soy phytoestrogen, and vitamin E (α -tocopherol succinate) (VitE) have a potent growth inhibitory effect on three cell lines derived from metastatic prostate adenocarcinomas: LNCaP, DU-145, and PC-3. In this study, we have examined the effects of these compounds on the levels of CB, CL and CPI both intracellularly and secreted into the medium. We find that after exposure to these compounds the levels of CB and CL drop below the level of detection. CPI levels, while falling dramatically, remain detectable and are found to have the same specific activity (% protease inhibition/mg protein) as in growing cells.

Examination of the stability of the acidic domain of N-arginine dibasic convertase, an opioid specific peptidase. KELLI CARPENTER* and EVA CSUHAI, Department of Chemistry, Transylvania University, 300 North Broadway, Lexington, KY 40508.

N-arginine dibasic convertase (NRDc) is a metallopeptidase. NRDc has been cloned and sequenced and has been shown to contain an unusually large number of acidic residues. It has been suggested that polyamines may regulate the activity of NRDc by binding to acidic residues located at the anionic domain of the enzyme [Csuhai *et al.* (1998) *Biochemistry* 37, 3787–3794]. The acidic domain of NRDc was produced and purified as a GST fusion protein in these experiments in order to run stability tests in the future by placing the fusion protein in a variety of media. The GST fusion proteins containing the acidic domain of mouse NRDc or human NRDc, as well as a control containing GST alone were scanned in a Circular Dichroism Spectrophotometer with and without the polyamine spermine in order to determine any structural effects spermine may have on the enzyme. There was no significant change in the percentage of alpha-helix, beta-turn, beta-sheet, or random portions in the enzyme. Therefore, spermine does not appear to have any significant effect on the secondary structure of the fragment containing the acidic domain of NRDc. Mouse NRDc was scanned in a Circular Dichroism Spectrophotometer with a temperature gradient in order to determine the effect that spermine has on the enzyme's heat stability. A concentration of 1 mM of spermine was used in the tests. There was no distinct heat transition observed under these conditions. Therefore, based on these results, spermine does not appear to affect the structure and stability of the acidic domain of NRDc.

SCIENCE EDUCATION

Classroom cheating: A survey. JOHN G. SHIBER, Prestonsburg Community College, Prestonsburg, KY 41653.

A survey of 877 high school students from four eastern Kentucky counties was conducted to determine the stu-

dents' attitude toward classroom cheating. Results: 38% said cheating in school is alright; 62% said it is wrong but 82% have done it; 95% have witnessed it; 67% would let friends copy their test answers if asked; 81% agreed plagiarism is cheating, and 56% admitted having done it, although 34% didn't know at the time they were plagiarizing; 33% do not regard copying friends' homework as cheating; 65% believe teachers cheat when they give grades higher or lower than students earn; 41% have been given a higher grade than they deserved at one time or other but only half brought it to the teacher's attention; and 47% have noticed teachers overlooking cheating, and, of these, 41% attribute it to teachers not caring what students do, 35% to favoritism, and 24% to their not wanting to embarrass the student or their desire to help the student pass the course. Compared to responses of 630 community college students to the same survey questions (Shiber 1999), those reported here suggest that high schoolers have a far more liberal attitude towards cheating and much greater experience with it than their older counterparts. The two groups' ideas about why students cheat are similar, however, with laziness to do work ranking first, lack of study time due to extra-curricular activities or family/job obligations second, too much pressure on good grades third, ineffective test-monitoring fourth, and, finally, many said students cheat because everybody cheats.

Megalitter: An Appalachian deformity. J. SMITH* and J. SHIBER, Prestonsburg Community College, Prestonsburg, KY 41653.

Three designated points, located on or near the banks of eastern Kentucky's Paintsville Lake, were systematically investigated for the first time for the presence, type, and abundance of megalitter over a 2-month period. Megalitter was collected by hand from each site every 2 weeks in March/ April 1999. It was sorted according to the type of material it was made from, counted, and recorded. By far, the most abundant type of megalitter was plastic, then styrofoam, paper/clothing, metals, and glass. As the weather warmed and more people frequented the park, the occurrence of megalitter tripled at the three locations. The incidence of plastic more than quadrupled. This pilot study indicates that a severe megalitter problem continues to exist within the environs of Paintsville Lake Park, despite the annual community clean-ups and daily efforts of the park personnel. The seriousness of illegal dumping and littering, not only in this park, but all over eastern Kentucky and other regions of the state, as witnessed by volunteers in the government-funded P.R.I.D.E. Program, makes it imperative that a mandatory educational program on waste management be implemented by the Commonwealth in the earliest stages of public schooling and maintained within the science curriculum throughout high school. Furthermore, appropriate penalties for littering and dumping can only be truly effective if more manpower is provided to enforce them, both in our public recreation facilities and the outlying communities.

ZOOLOGY & ENTOMOLOGY

The effects of pH levels on diversity and density of aquatic microorganisms. CHRIS ALTMAN and POLLY FROSTMAN,* Department of Biology, Transylvania University, Lexington, KY 40508.

To look at the potential effects of acid rain, we designed an experiment to determine whether or not there is a significant correlation between pH levels and diversity and/or density of aquatic microorganisms. Water was collected from a man-made lake and divided into nine equal samples. Each sample was examined to determine density and diversity under normal pH conditions. Then 20 ml of each sample (1–9) were removed. Half of these samples had their pH lowered to 6–6.5 and the other half had their pH lowered to 4 by adding aqueous sulfuric acid (H₂SO₄). Then the density and diversity of microorganisms present in each acidified sample was examined. Each set of data showed significant differences between the control samples and the acidified samples suggesting that the pH levels do affect the diversity and density of aquatic microorganisms. The pH levels used in this experiment are comparable to levels that could be caused by acid rain suggesting that acid rain can overcome the buffering capacity of water and affect aquatic ecosystems.

Optimal foraging in chickens (*Gallus domesticus*). SUMEET R. BHATT and BRIAN A. CAUDILL,* Department of Biology, Transylvania University, Lexington, KY, 40508.

We conducted a series of experiments to determine whether domesticated chickens (*Gallus domesticus*) were able to forage optimally under three different feeding conditions. Our study was conducted on three barnyard roosters and 10 hens fed at two feeding stations 3 m apart. In our first experiment, the chickens were presented with three times as much food at one feeding station as at the other. In the second experiment, chickens were fed twice as fast at one station as at the other. And in the third experiment, whole corn kernels were fed at one station, and an equivalent number of half corn kernels were fed at the other. The chickens showed a significant preference for feeding at the high quantity feeding station (first experiment) and the high feeding rate station (second experiment), but showed no significant preference for large food items over small (third experiment). This suggests that chickens are capable of making some basic decisions about where to forage to maximize their energy intake per unit time, although they did not seem very capable at discriminating between different sizes of food items that were presented in equal quantities.

Effects of continual food restriction on reproductive development and body organs in male house mice (*Mus musculus*). MICHAEL B. BOONE* and TERRY L. DERTING, Department of Biological Sciences, Murray State University, Murray, KY 42071.

Prior research has shown that moderate levels of intermittent food restriction have no negative effect on the reproductive development of male house mice. We determined the effects of moderate but continual food restriction on de-

velopment of the reproductive and digestive systems in post-weaning male house mice. We tested the null hypothesis that continual food restriction does not affect the level of testosterone or the masses of reproductive and body organs. Food intake of post-weaning males was restricted daily in a manner that prevented growth in overall body mass of the male. The males were sacrificed after 21 d and their level of testosterone and the wet and dry masses of their reproductive and other body organs were measured and compared with similar data from control males. The wet and dry masses of the testes were significantly lower in the food-restricted males. In contrast, testosterone levels of the food-restricted males were 55% higher, but not significantly different, compared with control males. The wet and dry mass of the stomach, but not the cecum, small intestine, and colon, was significantly heavier in food-restricted males compared with control males. Our results, in combination with those of studies using intermittent food restriction, indicate that when food resources are limited energy is allocated preferentially to processes necessary for reproduction (e.g., testosterone production for spermatogenesis). A negative impact of food restriction on reproductive processes in mammals may only occur when food restriction is severe.

Richness of terrestrial vertebrate species in Kentucky. MATTHEW L. COLE,* TERRY L. DERTING, and HOWARD WHITEMAN, Department of Biological Sciences, Murray State University, Murray, KY 40271.

To explore the diversity of terrestrial vertebrate species in Kentucky, we examined the distribution of species using richness (number of species) as an index of diversity. We evaluated species richness for amphibians, breeding birds, mammals, reptiles, and all terrestrial vertebrates combined using two different regional delineations from the US Forest Service: ecoregions and physiographic provinces. Using species ranges obtained from the scientific literature, species richness was calculated for each pixel (600 × 600 m) across the state. We overlaid the species richness maps with coverages of the ecoregions and provinces of Kentucky using a geographic information system (GIS). Among the ecoregions and provinces, mean species richness differed most for reptiles, particularly among the squamates. Mean species richness was highest for reptiles in the Mississippi Embayment and lowest in the Cumberland Mountains, a difference of 78%. In contrast, mean species richness was highest for amphibians and mammals in the Cumberland Mountains and lowest in the Bluegrass Region, a difference of approximately 20%. The variation in mean species richness for breeding birds was minimal, differing by no more than 10% among the physiographic provinces. Largely due to the high number of reptile species, mean species richness was 10–15% higher in the westernmost ecoregion and provinces compared with other geographic areas. Areas of highest species richness of terrestrial vertebrates were the Mississippi Embayment, Mississippi Alluvial Basin, and Cumberland Mountains. Management plans that protect these “hotspots” of species diversity will be necessary if the biodiversity of vertebrates in Kentucky is to be maintained.

List of Reviewers for Volume 61 of *Journal of the Kentucky Academy of Science*

David M. Brandenburg	Larry A. Giesmann	Thomas C. Rambo
Laura Burford	Stanley Hedeon	Patrick Schultheis
Jerry Carpenter	Miriam S. Kannan	Thomas Sproat
Sunni L. Carr	Laurie Lawrence	Karl Vogler
Ronald R. Cicerello	Barney Lipscomb	Mel L. Warren Jr.
Maria Falbo	James O. Luken	Gordon K. Weddle
Blaine Ferrell	Brainerd Palmer-Ball	Gary L. Uglem
Michael A. Flannery	Debra K. Pearce	George Yatskievych

INDEX TO VOLUME 61

Compiled by Varley Wiedeman

- Abies fraseri*, 52
Abietinella abietina, 117
 ABSTRACTS FROM 1999 KAS MEETING, 165–172
Acadian, 87
Accipiter striatus, 127
Acentrella ampla, 20
Acer pensylvanicum, 54
A. rubrum, 11, 54
A. saccharum, 11, 54
A. spicatum, 117
Achalarus lyciades, 86
Acipenser fulvescens, 125
Acleris youngana, 106
Aconitum uncinatum, 117
Acornshell, 130
Acorus calamus, 23, 26, 27
Acrobasis vaccinii, 106
Acroneuria, 14, 15
A. caroliniensis, 20
Actinastrum gracillimum, 36, 37
Actitis macularia, 127
 Adams cave beetle, lesser, 125
 ADAMS, KELLY, 62, 99
Adiantum capillus-veneris, 117
Adlumia fungosa, 117
Admiral
 red, 87
 white, 87
Aeschnidae, 21
Aesculus octandra, 11
A. pavia, 117
Agalinis auriculata, 117
A. obtusifolia, 117
A. skinneriana, 117
Agalis milberti, 87
Agastache scrophulariifolia, 117
Ageaius phoeniceus, 164
Ageratina altissima, 167, 168
 inheritance of achene characteristics in, 168
A. luciae-brauniae, 117
Agraulis vanillae, 87
 Agricultural sciences, 165–167
Agrimonia, 146–162
 in Kentucky, 146–162
A. eupatoria, 144, 147, 154, 155, 157–159
A. eupatoria × *A. procera*, 157
A. gryposepala, 117, 146, 147, 150, 151–154, 157, 158
A. incisa, 156
A. microcarpa, 146, 154, 155, 157
A. nipponica × *A. pilosa* var. *japonica*, 157
A. parviflora, 146–148, 150, 151–158
A. pilosa, 157, 158
A. pubescens, 146, 147, 150, 151–153, 157
A. rostellata, 146, 147, 150, 151–153
A. striata, 146, 155, 157, 158
A. suaveolens, 146
A. sylvatica, 146
Agropyron spicatum, 92
Aimophila aestivalis, 127
 Air breathing insects, control of, 166–167
Alabama lip fern, 118
Alabama shad, 125
Alasmidonta marginata, 124
A. atropurpurea, 124
 Algae, of Land Between the Lakes, 34–45
Allegheny chinkapin, 118
Allegheny stonecrop, 122
Alligator gar, 125
Alligator snapping turtle, 127
Allocapnia sp., 20
Alloperla sp., 20
Alosa alabamiae, 125
 ALTMAN, CHRIS, 171
Amblyopsis spelaea, 125
Amblyscirtes aesculapius, 86
A. belli, 86
A. hegon, 86
A. vialis, 86
Amelanchier laevis, 54
Ameletidae, 20
Ameletus, 15
A. sp., 20
American barberry, 117
American bison, 130
American bittern, 127
American brook trout, 126
American burying beetle, 125
American chaffseed, 122
American chestnut, 52, 118
American coot, 127
American copper, 87
American cow-wheat, 120
American crow, 53, 54
American crow-wheat, 120
American frog's-bit, 120
American golden-saxifrage, 118
American goldfinch, 56
American lady, 87
American lily-of-the-valley, 118
American redstart, 56
American snout butterfly, 87
American speedwell, 123
American water-pennywort, 119
American wintergreen, 121
Amianthium muscitoxicum, 117
Ammi majus, 159
Ammocrypta clara, 125
A. vivax, 130
Ammodramus henslowii, 127
Amphibians, 126–127
Amphinemura, 15
A. delosa, 20
Amphipod, 125
Amphipod, Bousfield's, 124
Amphiuma, three-toed, 126
Amphiuma tridactylum, 126
Amsonia tabernaemontana var. *gattingeri*, 117
Anaea andria, 87
Anartia jatrophae, 87
Anas clypeata, 127
A. discors, 127
Anatrytone logan, 86
Ancylid, domed, 123
Ancyloxipha numitor, 86
Anemone, Canada, 117
Anemone canadensis, 117
Angelica, filmy, 117
Angelica triquinata, 117
Angled riffleshell, 130
Anglepod, Carolina, 120
Anguispira rugoderma, 123
Anhinga, 130
Anhinga anhinga, 130
 Animal source oils, 166–167
 effectiveness on air breathing insects, 166–167
 Animals, 123–128, 130
Ankistrodesmus falcatus, 34, 35
A. spiralis, 36, 37
Anodontoides denigratus, 124
Anomodon rugelii, 117
Antaeotricha osseola, 105
Anthocharis midea, 87
Anthomyia, 155
 ANTONIOUS, GEORGE F., 23, 165
Antroselatus spiralis, 123
Apalone mutica mutica, 127
Apamea, undescribed species, 107

- Aphrodite fritillary, 87
 Apidae, 155
 Apioblasma haysiana, 130
 Apios priceana, 117
 Apis, 155
 Aporrectodea, 3
 Appalachian blue, 87
 Appalachian brown, 87
 Appalachian bugbane, 118
 Appalachian grizzled skipper, 125
 Appalachian rosinweed, 122
 Appalachian sandwort, 120
 Appalachian sedge, 118
 Appalachina chillhoweensis, 123
 Aquatic microorganisms, 171
 density, 171
 diversity, 171
 effects of pH levels on, 171
 Arabis hirsuta var. adpressipilis, 117
 A. missouriensis, 117
 A. perstellata, 117
 Archilochus colubris, 53
 Ardea alba, 127
 A. herodias, 127
 Aristida ramosissima, 117
 Armoracia lacustris, 117
 Armored rocksnail, 123
 Arrow head, 27
 Arrow-wood, Missouri, 123
 Arrowhead, delta, 122
 grass-leaf, 122
 sessile-fruit, 122
 Arrowwood, downy, 123
 Arthrodesmus convergens, 40, 41
 A. extensus, 40, 41
 A. octocornis, 40, 41
 Artificial nest density, 46–49
 effect on Canada Goose, 46–49
 Ascia, 155
 Asellidae, 22
 Ashcamp cave beetle, 127
 Ashy darter, 70, 126
 Asimina triloba, 163, 164
 Asio flammeus, 127
 A. otus, 127
 Aster
 barrens silky, 117
 eastern silvery, 117
 low rough, 117
 Rockcastle, 117
 Tennessee, 117
 Texas, 117
 white heath, 117
 whorled, 117
 Aster acuminatus, 117
 A. concolor, 117
 A. drummondii var. texanus, 117
 A. hemisphericus, 117
 A. pilosus var. priceae, 117
 A. pratensis, 117
 A. radula, 117
 A. saxicastellii, 117
 Asterocampa celtis, 87
 A. clyton, 87
 Atalopedes campestris, 86
 Atlides halesus, 87
 Atractosteus spatula, 125
 Atrichapogon sp., 21
 Atrytonopsis hianna, 86
 Auditory system model, 169
 pre-emphasis filter in, 169
 Aureolaria patula, 117
 Autochton cellus, 86
 Azalea, hoary, 121
 Azure
 dusky, 87
 spring, 87
 Azygiidae, 60–62, 99–104

 Baby-blue-eyes, small-flower, 121
 Bachman's sparrow, 127
 Bachman's warbler, 130
 Baetidae, 20
 Baetis sp., 20
 B. flavistriga, 20
 B. intercalaris, 15, 20
 B. tricaudatus, 15, 20
 BAIRD, NANCY DISHER, 83
 Bald eagle, 127
 Baltimore checkerspot, 87
 Bambusina brebissonii, 44, 45
 Banded darter, 70
 Banded 87
 Bank swallow, 128
 Baptisia australis var. minor, 117
 B. bracteata var. leucophaea, 117
 B. tinctoria, 117
 Barbara's-buttons, 120
 Barbed rattlesnake-root, 121
 Barberry, American, 117
 Barbicambarus cornatus, 124
 Barking treefrog, 127
 BARKSDALE, JAMES B., JR., 170
 Barn owl, 128
 feeding habits, 163–164
 Barrens silky aster, 117
 Bartonina virginica, 117
 Bartramia longicauda, 127
 Bashful bulrush, 122
 Bass
 hybrid striped, 166
 sunshine, 166
 diets for, 166
 Basswood, 54
 Bat
 evening, 128
 Rafinesque's big-eared, 128
 Virginia big-eared, 128
 Battus philenor, 86
 B. polydamas, 87
 Bay starvine, 122
 Beak-rush, woodland, 122
 Beaked-rush
 globe, 121
 tall, 121
 Bean
 Cumberland, 124
 rayed, 124
 Bear, black, 128
 Bearded skeleton, 119
 Beaver cave beetle, 125
 BEBE, F. N., 108
 Beebalm, spotted, 120
 Beetle
 American burying, 125
 Ashcamp cave, 125
 beaver cave, 125
 bold cave, 125
 cave, 125
 Clifton cave, 125
 concealed cave, 125
 Cub Run Cave, 125
 Garman's cave, 125
 Greater Adams Cave, 125
 hidden cave, 125
 icebox cave, 125
 lesser Adams cave, 125
 limestone cave, 125
 Louisville cave, 125
 Old Well Cave, 125
 Roger's cave, 125
 round-headed cave, 125
 scholarly cave, 12
 sixbanded longhorn, 125
 Stevens Creek Cave, 125
 surprising cave, 125
 Tatum Cave, Little Black Mountain, 50–59
 Bird-voiced treefrog, 126
 Birds, breeding, 127–128, 130
 Bishop's-weed
 eastern mock, 121
 mock, 121
 Nuttall's mock, 121
 Bison, American, 130
 Bittern
 American, 127
 least, 127
 Bivalva, 20
 Black bear, 128
 Black buffalo, 126
 Black locust, 54, 56
 Black lordithon rove beetle, 125
 Black swallowtail, 87
 Black tern, 130
 Black-and-white warbler, 56
 Black-crowned night-heron, 127
 Black-throated blue warbler, 54–56
 Blackberry, smooth, 122
 Blackbird, red-winged, 164
 Blackburnian warbler, 50, 53, 55, 127
 Blackfin sucker, 126
 Blackfoot quillwort, 120

- Blackside dace, 126
 Blacktail redhorse, 126
 Blacktail shiner, 125
 Bladderpod
 Lescur's, 120
 Lesquereux's, 120
 Bladderwort, greater, 123
 Bladetooth, Virginia, 123
 Blarina sp., 163
 B. brevicaudad, 164
 Blazingstar, slender, 120
 Bleufer, 124
 Bloodfin darter, 70
 Blossom
 tubercled, 130
 yellow, 130
 Blotched chub, 126
 Blotchside logperch, 130
 Blue grass, drooping, 121
 Blue heron
 great, 127
 little, 127
 Blue jasmine leather-flower, 118
 Blue jay, 54
 Blue monkshood, 117
 Blue mud-plantain, 117
 Blue scorpion-weed, 121
 Blue water I., 23, 26, 27
 Blue wild indigo, 117
 Blue
 Appalachian, 87
 eastern tailed, 87
 marine, 87
 silvery, 87
 Blue-flower coyote-thistle, 119
 Blue-headed vireo, 55
 Blue-joint reed 118
 Blue-star, eastern, 117
 Blue-winged teal, 127
 Blue-winged warbler, 55
 Bluebreast darter, 70
 Bluecurls, narrow-leaved, 123
 Bluegill, 73
 Bluets
 clustered, 121
 Michaux's, 119
 Blunt mountain-mint, 121
 Blunt-lobe grapefern, 117
 Bluntface shiner, 125
 Bobolink, 127
 Bog club-moss
 northern, 120
 southern, 120
 Bog goldenrod, southern, 122
 Bog lemming, southern, 163, 164
 Bog rush, 120
 Bog sedge
 brown, 118
 prickly, 118
 BOIADGIEVA, EMILIA 62, 99
 Bold cave beetle, 125
 Boloria bellona, 87
 B. selene myrina, 87
 Boluteloua curtispindula, 118
 Bombus, 155
 Bonasa umbellus, 53
 BONNEY, TERA M., 165, 166
 BOONE, MICHAEL B., 171
 Borer moth, rattlesnake-master, 125
 Bos bison, 130
 Botany & Microbiology, 167–168
 Botaurus lentiginosus, 127
 Botrychium matricariifolium, 117
 B. oneidense, 117
 Botryococcus braunii, 36, 37
 Bottlebrush crayfish, 124
 Bottomland lichen, 117
 Bousfield's amphipod, 124
 Boyeria vinosa, 21
 Boykinia aconitifolia, 118
 Brachythecium populeum, 117
 Branched three-awn 117
 Branta canadensis, 46
 Braun's rock-cress, 117
 Breeding birds, 127–128, 130
 Brighteye darter, 126
 Brilla sp., 22
 Bristly sedge, 118
 Broad-banded water snake, 127
 Broad-leaf golden-aster, 119
 Broad-winged skipper, 86
 Broadleaf water-milfoil, 121
 Broadwing sedge, 118
 Broken-dash
 northern, 86
 southern, 86
 Bronze copper, 87
 Brook lamprey
 mountain, 126
 northern, 126
 southern, 126
 Brook saxifrage, 118
 Brook snaketail, 125
 Brook trout, American, 126
 Broomrape, Louisiana, 121
 Brown bog sedge, 118
 Brown creeper, 127
 Brown elfin, 87
 Brown madtom, 126
 Brown, Appalachian, 87
 BROWN, D. KEVIN, 133
 BROWN, HETTI A., 164
 BROWN, SHAWN, 165
 Brown-headed cowbird, 50, 54, 56
 Bryocamptus morrisoni elegans, 124
 Bryum cyclophyllum, 117
 B. miniatum, 117
 Bubulcus ibis, 127
 Buckeye
 common, 87
 red, 117
 Buckley's goldenrod, 122
 Buffalo clover, 123–
 running, 123
 Buffalo, black, 126
 Bugbane, Appalachian, 118
 Bulbochaete varians, 36, 38
 Bull paspalum, 121
 Bulrush
 bashful, 122
 Hall's, 122
 river, 122
 slender, 122
 softstem, 23, 26, 27
 Bunchflower, Virginia, 120
 Bunting, indigo, 56
 Bur-reed, large, 122
 Burbot, 126
 Burhead, 119
 dwarf, 119
 Burnet, Canada, 122
 BURNS, A., 170
 BURRICHIO, T., 170
 Burrowing mayfly, 125
 robust pentagenian, 130
 Burying beetle, American, 125
 Bush's muhly, 120
 Bush-clover
 round-head, 120
 tall, 120
 Butler's quillwort, 120
 Buttercup, 27
 Butterflies, Kentucky, 86–87
 Butterfly, American snout, 87
 Button, wrinkled, 123

 Cabbage white, 87
 Cabomba caroliniana, 118
 Caddisflies, 15
 Helma's net-spinning, 125
 limnephilid, 125
 Cadmium, effect of on rats, 108–
 114
 Caecidotea sp., 22
 C. barri, 124
 Cajun dwarf crayfish, 124
 Calamagrostis canadensis var. ma-
 couniana, 118
 C. porteri ssp. insperata, 118
 C. porteri ssp. porteri, 118
 Calophelis borealis, 87
 C. mutica, 87
 Calla lily, 27
 Callirhoe alcaeoides, 118
 Callophrys augustuius, 87
 C. grynea, 87
 C. henrici, 87
 C. irus, 87
 C. nippon, 87
 Calopogon tuberosus, 118
 Calopterygidae, 21
 Calopteryx sp., 21
 Caloptilia fraxinella, 105
 Caltha palustris var. palustris, 130
 Calycanthus floridus var. glaucus,
 118
 Calycopis cecrops, 87
 Calylophus serrulatus, 118
 Cambarellus puer, 124
 C. shufeldtii, 124
 Cambaridae, 22
 Cambarus parvovulus, 124

- C. veteranus*, 124
 CAMPBELL, JULIAN J. N., 88
Campephilus principalis, 130
Campostoma anomalum, 75
Canada anemone, 117
Canada burnet, 122
Canada frostweed, 119
Canada goose, 46–49
 effect of artificial nest density on, 46–49
 effect of wetland size on, 46–49
 in constructed wetlands, 46–49
Canada warbler, 50, 53, 55, 128
Canadian yew, 123
 Canby's mountain-lover, 121
Canis lupus, 130
C. rufus, 130
Canola meal, 166
 in diets for fish, 166
Capniidae, 20
Caprifoliaceae, 30–33
Cardinal flower, 27
Cardinal, northern, 53, 56
Cardinalis cardinalis, 53
Carduelis tristis, 53, 56
Carex aestivalis, 118
C. alata, 118
C. appalachica, 118
C. atlantica ssp. *capillacea*, 118
C. austrocaroliniana, 118
C. buxbaumii, 118
C. comosa, 118
C. crawei, 118
C. crebriflora, 118
C. decomposita, 118
C. gigantea, 118
C. hystericina, 118
C. jorii, 118
C. juniperorum, 118
C. lanuginosa, 118
C. leptonervia, 118
C. reniformis, 118
C. roanensis, 118
C. rugosperma, 118
C. seorsa, 118
C. stipata var. *maxima*, 118
C. straminea, 118
C. tetanica, 118
Carolina anglepod, 120
Carolina fanwort, 118
Carolina larkspur, 119
Carolina parakeet, 130
Carolina satyr, 87
Carolina wren, 56
Carolina yellow-eye, 123
 CARPENTER, KELLI, 171
Carya spp., 11, 54
Carya aquatica, 118
Castanea dentata, 52, 118
C. pumila, 118
Castilleja coccinea, 118
 Catchfly
 ovate, 122
 royal, 122
Catharus fuscescens, 53, 55
 Catspaw, 124
 white, 130
 Cattails, 23–27
 Cattle egret, 127
 CAUDILL, BRIAN A., 171
 CAUDILL, TERESA L., 46
 Cave beetle, 125
 Ashcamp, 125
 beaver, 125
 bold, 125
 Clifton, 125
 concealed, 125
 Garman's, 125
 hidden, 125
 icebox, 125
 lesser Adams, 125
 limestone, 125
 Louisville, 125
 Roger's, 125
 round-headed, 125
 scholarly, 125
 surprising, 125
 Cave isopod, Clifton, 124
 Cavefish
 northern, 125
 southern, 126
 Cavesnail, shaggy, 123
Ceanothus herbaceus, 118
 Cedar sedge, 118
Celastrina argiolus ladon, 87
Celastrina ebenina, 87
C. neglectamajor, 87
Celephelis mutica, 125
Celithemis verna, 125
 Cellular & Molecular Biology, 168
 Central mudminnow, 126
 Central stoneroller, 75
Centropilum sp., 20
Ceratopogonidae, 21
Ceratopsyche sparna, 21
Cercyonis peagala, 87
Certhia americana, 127
Cerulean warbler, 50, 53
Cervus elaphus, 130
 Chaetophorales, 36
 Chaffseed, American, 122
 Chain pickerel, 126
Chalosyne gorgone, 87
 Channel darter, 70
Charadrius melodus, 116
Charidryas nycteis, 87
 Chat, yellow-breasted, 53
 Checkered white, 87
 Checkerspot
 Baltimore, 87
 Gorgone, 87
 silvery, 87
Cheilanthes alabamensis, 118
C. feei, 118
Chelone obliqua
 var. *obliqua*, 118
 var. *speciosa*, 118
 Chestnut lamprey, 126
 Chestnut, American, 52, 118
 Chestnut-sided warbler, 54, 55, 56
Cheumatopsyche sp., 21
C. helma, 125
 Chickens, optimal foraging in, 171
 Chinkapin, Allegheny, 118
Chionodes, undescribed species, 105
C. adamas, 106
C. aruns, 106
C. baro, 106
C. hapsus, 105
C. sevir, 106
C. suasor, 105
Chlidonias niger, 130
Chironomidae, 22
Chloralictus, 155
Chlorococcales, 34
Chlorogonium euchlorum, 34, 35
Chloroperlidae, 20
Chlorophyta, 34–45
Chondestes grammacus, 127
Chrysemys picta dorsalis, 127
Chrysogonium virginianum, 118
Chrysosplenium americanum, 118
 Chub
 blotched, 126
 flame, 130
 flathead, 126
 gravel, 130
 hornyhead, 126
 sicklefin, 126
 sturgeon, 126
 Chubsucker, lake, 126
Cimicifuga rubifolia, 118
Cinygmula, 15
C. subaequalis, 20
Circaea alpina, 118
Circus cyaneus, 127
Cirriphyllum piliferum, 117
Cistothorus platensis, 127
 CLARK, E. P., 169
 Classroom cheating, 171
 Cleft phlox, 121
 starry, 121
Clematis crispa, 118
 CLEMMONS, ANTON M., 167
Clethrionomys gapperi maurus, 128
 Clifton cave beetle, 125
 Clifton cave isopod, 124
 Clifty covert, 123
 Climbing fumitory, 117
Clioperla clio, 20
 Cloak, mourning, 87
Clonophis kirtlandii, 127
Closteriopsis longissima, 36, 37
Closterium abruptum, 36, 39
C. ehrenbergii, 36, 39
C. setaceum, 36, 39
 Clouded skipper, 86
 Clouded sulphur, 87
 Cloudless sulphur, 87
 Cloudywing
 confused, 86

- northern, 86
 southern, 86
 Clover
 buffalo, 123
 running buffalo, 123
 Club-moss
 northern bog, 120
 southern bog, 120
 Clubshell, 124
 Tennessee, 124
 Clubtail, elusive, 125
 Clustered bluets, 121
 Clustered poppy-mallow, 118
 Coachwhip, eastern, 130
 Coal skink
 northern, 127
 southern, 127
 Coastal Plain sedge, 118
 Cobweb skipper, 86
 Coccyzus americanus, 53
 Coelastrum cambricum, 36, 37
 C. microporum, 36, 37
 Coeloglossum viride var. virescens, 118
 Coil, punctate, 123
 Colaptes auratus, 53
 COLE, CHRISTY, 168
 COLE, MATTHEW L., 171
 Coleochaete orbicularis, 36, 38
 C. scutata, 36, 38
 Coleoptera, 21
 Colias cesonia, 87
 C. eurytheme, 87
 C. philodice, 87
 Collinsonia verticillata, 118
 Columbine duskywing, 86
 COMBS, MICHAEL S., 133
 Combshell
 Cumberlandian, 124
 round, 130
 Comma
 buckeye, 87
 checkered skipper, 86
 eastern, 87
 gray, 125
 green, 87, 125
 gray, 87
 Common moorhen, 127
 Common raven, 127
 Common roadside skipper, 86
 Common silverbell, 119
 Common sootywing, 86
 Common wood-nymph, 87
 Common yellowthroat, 53
 Compassplant, 122
 Compton tortoise shell, 87
 Comptonia peregrina, 118
 Concealed cave beetle, 125
 Conchapelopia sp., 22
 Coneflower, sweet, 122
 Confused cloudywing, 86
 Conjurer's-nut, 121
 Conradina verticillata, 118
 Constempellina sp., 22
 Constructed wetlands, 23–29
 Canada Goose in, 46–49
 Contopus virens, 53
 Conuropsis carolinensis, 130
 Convallaria montana, 118
 Coot, American, 127
 Copepod, 124
 Copper iris, 120
 Copper
 American, 87
 bronze, 87
 Copperbelly water snake, 127
 Coral, 87
 Corallorhiza maculata, 118
 Cordulegaster, 14
 C. sp., 21
 Cordulegasteridae, 21
 Coreopsis pubescens, 118
 Cormorant, double-crested, 128
 Corn snake, 127
 Corvus brachyrhynchos, 53
 C. corax, 127
 C. ossifragus, 127
 Corydalidae, 21
 Corydalis, pale 118
 Corydalis sempervirens, 118
 Corynoneura sp., 22
 Corynorhinus rafinesquii, 128
 C. townsendii virginianus, 128
 Cosmarium baileyi, 39, 40
 C. bipunctatum, 39, 40
 C. biretum, 39, 40
 C. blyttii, 39, 40
 C. botrytis, 39, 40
 C. depressum, 39, 40
 C. granatum, 39, 40
 C. margaritatum, 39, 40
 C. meneghinii, 40, 41
 C. moniliforme, 40, 41
 C. nymmannianum, 40, 41
 C. obtusatum, 40, 41
 C. orthostichum, 40, 41
 C. ovale, 40, 41
 C. phaseolus, 40, 41
 C. porrectum, 40, 41
 C. portianum, 40, 41
 C. pyramidatum, 40, 41
 C. subtumidum, 40, 41
 C. turpinii, 40, 41
 Cosmocladium pusillum, 40, 41
 Cotton mouse, 128
 Cotton- tawny, 119
 COVELL, CHARLES V., JR., 86, 105
 Covert, cliffty, 123
 Cow-parsnip, 119
 Cow-wheat, American, 120
 Cowbird, brown-headed, 50, 54, 56
 COYLE, SHAWN, 165, 166, 167
 Coyote-thistle, blue-flower, 119
 Craba cuneifolia, 119
 Crabapple, southern, 120
 Cracking pearlymussel, 130
 Crambidae, 105, 106
 Crater, queen, 123
 Crawe's sedge, 118
 Crawfish frog, northern, 127
 Crayfish, 124, 125
 bottlebrush, 124
 Cajun dwarf, 124
 Crittenden, 124
 dwarf, 124
 Louisville, 124
 Cream wild indigo, 117
 Creek heelsplitter, 124
 Creekshell
 Kentucky, 124
 mountain, 124
 Creeper, brown, 127
 Creeping St. John's-wort, 119
 Creole pearly-eye, 87
 Crescent
 pearl, 87
 tawny, 87, 125
 Cress
 glade, 120
 lake, 117
 necklace glade, 120
 Cricotopus trifascia, 22
 Crinkled hair 119
 Crittenden crayfish, 124
 Cross-leaf milkwort, 121
 Crossline skipper, 86
 Crow
 American, 53, 54
 fish, 127
 Crow-wheat, American, 120
 Crucigenia tetrapedia, 36, 37
 Crustacea, 22
 Crustaceans, 124–125
 Cryocopus pileatus, 53
 Cryptobranchus allaganiensis alle-ganiensis, 126
 Cryptotis parva, 164
 Crystal darter, 130
 Crystallaria asprella, 130
 CSUHAI, EVA, 171
 Cub Run Cave beetle, 125
 Cuckoo, yellow-billed, 53
 Cumberland bean, 124
 Cumberland elktoe, 124
 Cumberland leafshell, 130
 Cumberland papershell, 124
 Cumberland rosemary, 118
 Cumberland sandwort, 120
 Cumberlandia monodonta, 124
 Cumberlandian combshell, 124
 Cupped vertigo, 124
 Curtis' goldenrod, 122
 Cut turf, 167–168
 volatile emissions from, 167–168
 Cutleaf meadow-parsnip, 123
 Cutleaf water-milfoil, 121
 Cyanocitta cristata, 53, 54
 Cyllopsis gemma, 87
 Cymophyllus fraserianus, 118
 Cynodon dactylon, 167
 Cyperus plukenetii, 118

- Cyperus, Plukenet's, 118
 Cypress darter, 126
 Cypress minnow, 126
 Cypress-swamp sedge, 118
 Cyprinella camura, 125
 C. venusta, 125
 Cypripedium candidum, 118
 C. kentuckiense, 118
 C. parviflorum, 119
 C. reginae, 119
 Cyprogenia stegaria, 124
- Dace
 blackside, 126
 longnose, 126
 Dainty sulphur, 87
 Dalea purpurea, 119
 Danaus gilippus, 87
 D. plexippus, 87
 Dandelion, 167
 western dwarf, 120
 DARIA, D., 170
 Dark-eyed junco, 50, 53, 55, 127
 Darter
 ashy, 70, 126
 banded, 70
 bloodfin, 70
 bluebreast, 70
 brighteye, 126
 channel, 70
 crystal, 130
 cypress, 126
 duskytail, 67–76, 126
 emerald, 70
 firebelly, 126
 goldstripe, 126
 greenside, 70
 gulf, 126
 johnny, 126
 least, 130
 longhead, 126
 olive, 126
 rainbow, 70
 relict, 126
 scaly sand, 130
 Shawnee, 126
 smallscale, 126
 speckled, 70
 spotted, 126
 swamp, 126
 tippecanoe, 70
 western sand, 125
 Darters, 60
 DAVIS, MARIA L., 167
 Decapoda, 22
 Delaware skipper, 86
 Delicate vertigo, 124
 Delphinium carolinianum, 119
 Delta arrowhead, 122
 Dendroica caerulescens, 53, 54
 D. cerulea, 50, 53
 D. coronata, 53
 D. fusca, 50, 53, 127
 D. pensylvanica, 53, 54
 D. tigrina, 53
 D. virens, 53
 Dero nivea, 20
 DERTING, TERRY L., 171
 Deschampsia cespitosa ssp. glauca, 119
 D. flexuosa, 119
 Desmidium aptogonum, 44
 D. grevillii, 44
 D. swartzii, 44, 45
 Dewberry, Wharton's, 122
 Diamesa sp., 22
 Diana fritillary, 87
 Diasemodes nigralis, 106
 Dichanthelium boreale, 119
 Dicranodontium asperulum, 117
 Dicranota sp., 22
 Dictyosphaerium pulchellum, 36, 37
 Didiplis diandra, 119
 Digenea, 99–104
 Digenetic trematode, 60–63
 Digital model
 analysis of, 169
 of mammalian basilar membrane, 169
 DILLARD, GARY E., 34
 Dion skipper, 86
 Diplectrona, 10, 13, 15
 D. modesta, 21
 Diplocadius sp., 22
 Diploperla robusta, 20
 Diptera, 21, 155
 Disporum maculatum, 119
 Dixa sp., 22
 Dixidae, 22
 Docidium baculum, 36, 39
 D. undulatum, 36, 39
 Dodecatheon frenchii, 119
 Dogface, southern, 87
 Dolichonyx oryzivorus, 127
 Dollar sunfish, 126
 Domed ancyrid, 123
 Double-crested cormorant, 128
 Double-ringed pennant, 125
 Dove, mourning, 53
 Downy arrowwood, 123
 Downy goldenrod, 122
 Dragon-head, slender, 130
 Dreamy duskywing, 86
 Dromedary pearlymussel, 130
 Dromus dromas, 130
 Drooping blue, 121
 Dropseed
 northern, 122
 rough, 122
 Drosera brevifolia, 119
 D. intermedia, 119
 Dryobius sexnotatus, 125
 Dryopidae, 21
 Dryopteris carthusiana, 119
 D. ludoviciana, 119
 Duke's skipper, 86, 125
 Dun skipper, 86
 Dusky azure, 87
 Duskytail darter, 67–76, 126
 Duskywing
 columbine, 86
 dreamy, 86
 funereal, 86
 Horace's, 86
 Juvenal's, 86
 mottled, 86
 sleepy, 86
 wild indigo, 86
 zarucco, 86
 Dusted skipper, 86
 Dwarf burhead, 119
 Dwarf crayfish, 124
 Cajun, 124
 Dwarf dandelion, western, 120
 Dwarf sundew, 119
- Eagle, bald, 127
 Ealophrys irus, 125
 Earleaf false foxglove, 117
 Early 87, 125
 Earthworms, 1–5
 Eastern blue-star, 117
 Eastern coachwhip, 130
 Eastern comma, 87
 Eastern eulophus, 121
 Eastern hellbender, 126
 Eastern mock bishop's-weed, 121
 Eastern phoebe, 53
 Eastern pine elfin, 87
 Eastern puma, 130
 Eastern ribbon snake, 127
 Eastern silvery aster, 117
 Eastern slender glass lizard, 127
 Eastern small-footed myotis, 128
 Eastern spotted skunk, 128
 Eastern tailed blue, 87
 Eastern turkeybeard, 123
 Echinodorus berteroi, 119
 E. parvulus, 119
 Eclipidrilus sp., 20
 Ectopistes migratorius, 130
 Ectopria, 15
 E. nervosa, 21
 Edward's, 87
 Eel-grass, 123
 Eggert's sunflower, 119
 Eggleston's violet, 123
 Egret
 cattle, 127
 great, 127
 Egretta carulea, 127
 EISENHOUR, DAVID J., 67
 Eiseniella, 3
 Elanoides forficatus forficatus, 130
 Elaphe guttata guttata, 127
 Elaphria cornutinis, 107
 E. festivoides, 107
 Eleocharis olivacea, 119
 Elfin
 brown, 87
 eastern pine, 87
 frosted, 87

- Henry's, 87
Elimia ebumum, 60–63
E. semicarinata, 60, 61
 Elk, 130
 Elktoe, 124
 Cumberland, 124
 Elm, September, 123
 Elmidae, 21
Elodea nuttallii, 119
Elusive clubtail, 125
Elymus species, 88–98
E. elymoides, 88
E. glabriflorus, 88, 89, 92
E. glaucus, 88, 92, 93
 ssp. *glaucus*, 96
 ssp. *jepsonii*, 93, 96
 ssp. *mackenzii* comb nov., 88–98
 ssp. *virescens*, 93
 var. *minor*, 88
E. macgregorii sp. nov., 88–98
E. mackenzii, 93
E. stebbinsii, 92
E. svenssonii, 119
E. trachycaulus, 93, 95
E. villosus, 92
E. virginicus, 88, 89, 92
 var. *glabriflorus*, 92
 var. *intermedius*, 89, 92
 var. *jejunus*, 88, 89, 92
 var. *minor*, 92
 Emerald darter, 70
 Emperor
 hackberry, 87
 tawny, 87
 Empididae, 22
Empidonax minimus, 127
E. virescens, 53
 Enchanter's-nightshade, small, 118
Endopiza yaracana, 106
Enodia anthedon, 87
E. creola, 87
E. portlandia missarkae, 87
Entodon brevisetus, 117
Epargyreus clarus, 86
Epeorus, 10, 15
E. prob. namatus, 20
Ephemera guttulata, 20
E. simulans, 20
Ephemerella, 10, 15
E. inconstans, 125
E. prob. auravilii, 20
Ephemerellid mayfly, 125
 Ephemerellidae, 20
 Ephemeridae, 20
 Ephemeroptera, 20
Epioblasma arcaeiformis, 130
E. biemarginata, 130
E. brevidens, 124
E. capsaeformis, 124
E. flexuosa, 130
E. florentina florentina, 130
E. florentina walkeri, 130
E. lewisii, 130
E. obliquata obliquata, 124
E. obliquata perobliqua, 130
E. personata, 130
E. propinqua, 130
E. sampsonii, 130
E. stewardsonii, 130
E. torulosa rangiana, 124
E. torulosa torulosa, 130
E. triquetra, 124
 Epiphytic sedge, 118
 Epling's hedge-nettle, 122
Eremosphaera viridis, 34, 35
Erimystax insignis, 126
Erimystax x-punctatus, 130
Erimyzon sucetta, 126
Eriophorum virginicum, 119
Eristalis, 155
Erora laetus, 87, 125
Eryngium intergrifolium, 119
Erynnis baptisiae, 86
Erynnis brizo, 86
E. funeralis, 86
E. horatius, 86
E. icelus, 86
E. juvenalis, 86
E. lucilius, 86
E. martialis, 86
E. zarucco, 86
Erythronium rostratum, 119
Esox lucius, 75
E. niger, 126
Etheostoma spp., 60
E. baileyi, 70
E. blennioides, 70
E. caeruleum, 70
E. camurum, 70
E. chienense, 126
E. cinereum, 70, 126
E. flabellare, 67, 68
E. fusiforme, 126
E. lynceum, 126
E. maculatum, 126
E. microlepidum, 126
E. microperca, 130
E. nigrum susanae, 126
E. parvipinne, 126
E. percunum, 67–76, 126
E. proeliare, 126
E. pyrrhogaster, 126
E. sanguifluum, 70
E. stigmaeum, 70
E. swaini, 126
E. tecumsehi, 126
E. tippecanoe, 70
E. virgatum, 60, 61
E. zonale, 70
Euastrum abruptum, 40, 43
E. affine, 40, 43
E. ansatum, 40, 43
E. binale, 40, 43
E. denticulatum, 40, 43
E. didelta, 40, 43
E. elegans, 40, 43
E. evolutum, 40, 43
E. insulare, 40, 43
E. verrucosum, 40, 43
Euchloe olympia, 87
Eufala skipper, 86
Eukiefferiella spp., 22
Eulophus, eastern, 121
Eumeces anthracinus anthracinus, 127
E. anthracinus pluvialis, 127
E. inexpectatus, 127
Eupatorium maculatum, 119
E. rugosum, 167
E. semiserratum, 119
E. steelei, 119
Euphorbia mercurialina, 119
Euphydryas phaeton, 87
Euphyes dion, 86
E. dukesi, 86, 125
E. vestris, 86
Euptoieta claudia, 87
Eurema lisa, 87
E. nicippe, 87
 European skipper, 86
Eurycea guttolineata, 126
Eurylophella funeralis, 20
Eurytides marcellus, 87
 EVEN, DEBORAH, 168
 Evening bat, 128
 Evening primrose, 121
 yellow, 118
 stemless, 121
Everes comyntas, 87
 Extirpated biota of Kentucky, 115–132

Fagus grandifolia, 11
 Falcate Orange Tip, 87
Falco peregrinus, 127
 Falcon, peregrine, 127
 False foxglove
 earleaf, 117
 pale, 117
 spreading, 117
 ten-lobed, 117
 False gromwell
 hairy, 121
 soft, 121
 western, 121
 False hellebore, 120
 small-flowered, 120
 False mallow, hispid, 120
 False solomon-seal, starry, 120
 Fameflower
 limestone, 122
 roundleaf, 123
 Fanshell, 124
 Fanwort, Carolina, 118
Farancia abacura reinwardtii, 127
 Fat pocketbook, 124
 Fecal coliform species, 168
 from Lee's Branch, 168
 identification of, 168
Fèe's lip fern, 118
Feniseca tarquinius, 87
 Fern

- Alabama lip, 118
 Fee's lip, 118
 southern maidenhair, 117
 southern shield wood, 119
 spinulose wood, 119
FERRELL, SHELLY, 169
 Fescue, 56
Festuca arundinacea, 56
F. elatior, 27
 Fetterbush, 120
 Few-flowered scurf-pea, 121
 Fiddleleaf
 one-flower, 119
 ovate, 119
 Fiery skipper, 86
 Filmy angelica, 117
Fimbristylis, hairy, 119
Fimbristylis purberula, 119
 Finely-nerved sedge, 118
 Fir, Fraser, 52
 Firebelly darter, 126
 Fish crow, 127
 Fishes, 125–126, 130
 Five-lined skink, southeastern, 127
 Flame chub, 130
 Flathead chub, 126
 Floater, green, 124
 Fluted kidneyshell, 124
 Fly-poison, 117
 Flycatcher, least, 127
Forcipomyia sp., 21
Forestiera ligustrina, 119
 Forkshell, 130
 Fox grape, northern, 123
 Foxglove
 earleaf false, 117
 pale false, 117
 spreading false, 117
 ten-lobed false, 117
 Fraser fir, 52
 Fraser's loosestrife, 120
 Fraser's sedge, 118
Fraxinus americana, 54
 French's shooting-star, 119
FRENCH, AUSTIN, 170
 Freshwater mussels, 124, 130
 Freshwater prawns, 165, 166
 copepods as live food for, 165–166
 effect of water temperature on survival, 167
 transport density on survival, 165
 Fringed nut-rush, 122
 Fringeless orchid, white, 121
 Fritillary
 Aphrodite, 87
 Diana, 87
 great-spangled, 87
 gulf, 87
 meadow, 87
 regal, 87, 125
 silver-bordered, 87
 variegated, 87
FRITZ, D., 170
 Frog's-bit, American, 120
 Frog
 northern crawfish, 127
 northern leopard, 127
 Frosted elfin, 87, 125
FROSTMAN, POLLY, 171
 Frostweed
 Canada, 119
 plains, 119
Fulica americana, 127
 Fumitory, climbing, 117
Fumonelix wetherbyi, 123
Fundulus chrysotus, 126
F. dispar, 126
 Funereal duskywing, 86
Fusconaia subrotunda subrotunda, 124
 GABAc rho1 subunit genes, 168
 in the mouse, 168
 GABAc rho2 subunit genes, 168
 in the mouse, 168
Gallinula chloropus, 127
Gallus domesticus, 171
Gammarus bousfieldi, 124
 Gar, alligator, 125
 Garman's cave beetle, 125
 Gastropods, 123–124
 Gattinger's lobelia, 120
 Gaywings, 121
 Gelechiidae, 105
 Gemmed satyr, 87
 Genistein, 170–171
 effect on Cathepsin B, 170–171
 effect on Cathepsin L, 170–171
 effect on cysteine proteases, 170–171
 Gentian
 prairie, 119
 showy, 119
 yellow, 119
Gentiana decora, 119
G. flavida, 119
G. puberulenta, 119
 Geometrid moth, 125
 Geometridae, 105, 107
Geothlypis trichas, 53
 Gian hyssop, purple, 117
 Giant swallowtail, 87
GIBSON, LORAN D., 105
Gillmeria pallidactyla, 107
 Glade cress, 120
 necklace, 120
Glandularia canadensis, 119
 Glass lizard, eastern slender, 127
 Glassy grapeskin, 124
 Glassywing, little, 86
Glaucopsyche lygdamus, 87
Glechoma hederacea, 167
Gleditsia aquatica, 119
 Globe beaked-rush, 121
Gloeocystis planctonica, 34, 35
Glossosoma sp., 21
 Glossosomatidae, 21
Glyceria acutiflora, 119
 Glyph
 Maryland, 123
 sculpted, 123
Glyphyalinia raderi, 123
G. rhoadsi, 123
Gnaphalium helleri var. *micradenium*, 119
 Goatweed leafwing, 87
 Goera sp., 21
 Goeridae, 21
 Goerita, 15
G. betteni, 21
 Gold-banded skipper, 86
 Golden topminnow, 126
 Golden-aster, broad-leaf, 119
 Golden-crowned kinglet, 55
 Golden-saxifrage, American, 118
 Golden-star, 119
 Golden-winged warbler, 50, 53, 55, 128
 Goldenclub, 121
 Goldenrod
 Buckley's, 122
 Curtis', 122
 downy, 122
 Rand's, 122
 Roan Mountain, 122
 Short's, 122
 southern bog, 122
 squarrose, 122
 white-haired, 122
 Goldfinch, American, 56
 Goldstripe darter, 126
Golenkinia radiata, 36, 37
 Gomphidae, 21
Gonatozygon brebissonii, 36, 39
 Gorgone checkerspot, 87
GOTTESMAN, STEPHEN T., 170
 Gracillariidae, 105
 Grama, side-oats, 118
 Grape honeysuckle, 120
 Grape
 northern fox, 123
 sand, 123
 Grapefern
 blunt-lobed, 117
 matricary, 117
 Grapeskin, glassy, 124
 Grass
 bearded skeleton, 119
 blue-joint reed, 118
 branched three-awn, 117
 crinkled hair, 119
 drooping blue, 121
 hair, 120
 June, 120
 northern witch, 119
 pale manna, 123
 Porter's reed, 118
 purple sand, 123
 reed bent, 118
 reed canary, 23, 26, 27
 sharp-scaled manna, 119

- shortleaf skeleton, 119
 tufted hair, 119
 Grass-leaf arrowhead, 122
 Grass-of-parnassus
 kidney-leaf, 121
 largeleaf, 121
 Grass-pink, 118
 Grassleaf mud-plantain, 119
 Gratiola pilosa, 119
 G. viscidula, 119
 Gravel chub, 130
 Gray comma, 87, 125
 Gray hairstreak, 87
 Gray myotis, 128
 Gray treefrog, 127
 Gray wolf, 130
 Great blue heron, 127
 Great egret, 127
 Great Plains ladies'-tresses, 122
 Great purple, 87
 Great-spangled fritillary, 87
 Greater Adams Cave beetle, 125
 Greater bladderwort, 123
 Greater prairie-chicken, 130
 Greater redhorse, 130
 Grebe, pied-billed, 128
 Green comma, 87, 125
 Green floater, 124
 Green orchis, long-bract, 118
 Green treefrog, 127
 Green water snake, Mississippi, 127
 Green-and-gold, 118
 Greenside darter, 70
 GRISBY, EBONY J., 166
 Grizzled skipper, 86
 Appalachian, 125
 Gromwell
 hairy false, 121
 soft false, 121
 western false, 121
 Groovebur, tall hairy, 117
 Grosbeak, rose-breasted, 50, 53, 55,
 56, 128
 Ground juniper, 120
 Gulf darter, 126
 Gulf fritillary, 87
 Gymnopogon ambiguus, 119
 G. brevifolius, 119

 Hackberry emperor, 87
 Hair 120
 Hair
 crinkled, 119
 tufted, 119
 Haircap moss, 117
 Hairstreak
 Acadian, 87
 banded, 87
 coral, 87
 early, 87, 125
 Edward's, 87
 gray, 87
 great purple, 87
 hickory, 87
 juniper, 87
 northern, 87, 125
 red-banded, 87
 striped, 87
 white-m, 87
 Hairy false gromwell, 121
 Hairy fimbriatilis, 119
 Hairy groovebur, tall, 117
 Hairy hawkweed, 119
 Hairy ludwigia, 120
 Hairy rock-cress, 117
 Hairy skullcap, 122
 Hairy snout-bean, 121
 HAISCH, KARL E., JR., 170
 Halesia tetraptera, 119
 Haliaeetus leucocephalus, 127
 Halictidae, 155
 Halictus, 155
 Hall's bulrush, 122
 Haploperla sp., 20
 Haplotaxida, 20
 Harelip sucker, 130
 Harrier, northern, 127
 Harvest mouse, 163, 164
 Harvester, 87
 Hawk, sharp-shinned, 127
 Hawkweed, hairy, 119
 Hayhurst's scalloppwing, 86
 Health Sciences, 169–170
 Heartleaf plantain, 121
 Heartleaf
 southern, 119
 variable-leaved, 119
 Heath aster, white, 117
 HEDEEN, STANLEY E., 6
 Hedeoma hispidum, 119
 Hedge-hyssop
 shaggy, 119
 Short's, 119
 Hedge-nettle, Epling's, 122
 Heelsplitter, creek, 124
 Helianthemum bicknellii, 119
 H. canadense, 119
 Helianthus eggertii, 119
 H. silphioides, 119
 Helichus fastigiatus, 21
 Helicodiscus notius specus, 123
 H. puntatellus, 123
 Hellbender, eastern, 126
 Hellebore
 false, 120
 small-flowered false, 120
 Helma's net-spinning caddisfly, 125
 Helmitheros vermivorus, 53
 Helvibotys pseudohelvalis, 105
 Hemerodromia sp., 22
 Hemistena lata, 130
 Hemitremia flammea, 130
 Hempseed meal, 166
 in diets for fish, 166
 Henry's elfin, 87
 Henslow's sparrow, 127
 HENSON, G., 170
 Heptageniid mayfly, 125
 Heptageniidae, 20
 Heracleum lanatum, 119
 Hermeuptychia sosybius, 87
 HERNER-THOGMARTIN, JEN-
 NIFER H., 163
 Heron
 great blue, 127
 little blue, 127
 Herzogiella turfacea, 117
 Hesperia leonardus, 86
 H. metea, 86
 H. sassacus, 86
 Heteranthera dubia, 119
 H. limosa, 119
 Heterotheca subaxillaris var. latifol-
 ia, 119
 Hexastylis contracta, 119
 H. heterophylla, 119
 Hexatoma sp., 22
 Hickory, 87
 water, 118
 Hidden cave beetle, 125
 Hieracium longipilum, 119
 Hispid false mallow, 120
 Hoary azalea, 121
 Hoary edge, 86
 Hoary mock orange, 121
 Hoary-pea, spiked, 123
 Hobomok skipper, 86
 HOLDEN, T., 170
 Homoeosoma deceptorium, 107
 Honeysuckle
 grape, 120
 wild, 120
 Hooded merganser, 127
 Hooded warbler, 54–56
 Horace's duskywing, 86
 Hornsnail
 rugged, 123
 shortspire, 123
 Hornyhead chub, 126
 Horse-balm, whorled, 118
 House mouse, 164
 body organs, 171
 effect of food restriction, 171
 male, 171
 reproductive development, 171
 Houstonia serpyllifolia, 119
 HUGO, E., 170
 Hummingbird, ruby-throated, 53
 HURST, B., 170
 Hyalotheca dissiliens, 44
 H. mucosa, 44
 Hybognathus hayi, 126
 H. placitus, 126
 Hybopsis amnis, 126
 Hybrid striped bass, 166
 Hydrobaenus sp., 22
 Hydrocotyle americana, 119
 Hydrogen gas in shell galaxies, 170
 Hydrolea ovata, 119
 H. uniflora, 119
 Hydrophyllum virginianum, 119
 Hydropsyche betteni, 21

- Hydropsychidae, 21
Hyla avivoca, 126
H. cinerea, 127
H. gratiosa, 127
H. versicolor, 127
Hylephila phyleus, 86
Hylocichla mustelina, 53
H. mustelina, 56
 Hymenoptera, 155
Hypericum adpressum, 119
H. crux-andreae, 119
H. nudiflorum, 120
H. pseudomaculatum, 120
 Hyssop, purple giant, 117
- Icebox cave beetle, 125
Ichthyomyzon castaneus, 126
I. fossor, 126
I. gagei, 126
I. greeleyi, 126
Icteria virens, 53
Icterus spurius, 53
Ictinia mississippiensis, 127
Ictiobus niger, 126
 Illinois pondweed, 121
Immyria nigrovittella, 106
 Indian paintbrush, scarlet, 118
 Indian skipper, 86
 Indian wild rice, 123
 Indiana myotis, 128
 Indigo bunting, 56
 Indigo
 blue wild, 117
 cream wild, 117
 yellow wild, 117
 Inland silverside, 126
 Insecta, 20, 105–107
 Insects, 125, 130
 air breathing, control of, 166–167
Iris fulva, 120
I. pseudacorus, 27
I. versicolor, 23, 26, 27
 Iris
 blue water, 23, 26, 27
 copper, 120
 yellow water, 27
Ironoquia punctatissima, 21
 Ironweed, New York, 123
Isoetes butleri, 120
I. melanopoda, 120
Isoperla holochlora, 20
 Isopod, Clifton cave, 124
 Isopoda, 22
 Ivory-billed woodpecker, 130
Ixobrychus exilis, 127
- JAMES, MICHAEL A., 133
 Jay, blue, 54
 Joan's swallowtail, 87
 Joe-pye-weed
 spotted, 119
 Steele's, 119
 Johnny darter, 126
 Jointed rush, 120
- JONES, BRITTNEY, 1
 JONES, SNAKE C., 165, 166
 JUETT, BEVERLY W., 168
Juglans cinerea, 120
Junco hyemalis, 50
J. hyemalis, 53, 127
Junco, dark-eyed, 50, 53, 55, 127
Juncus articulatus, 120
J. elliotii, 120
J. filipendulus, 120
 June, 120
 Juniper, 87
 ground, 120
Juniperus communis var. *depressa*, 120
Junonia coenia, 87
 Juvenal's duskywing, 86
- KALISZ, PAUL J., 1
 Kentucky creekshell, 124
 Kentucky lady's-slipper, 118
 Kentucky red-backed vole, 128
 KENTUCKY STATE NATURE PRESERVES COMMISSION, 115
 Kentucky vertebrate species, 171
 Kidney-leaf grass-of-parnassus, 121
 Kidney-leaf twayblade, 120
 Kidneyshell, fluted, 124
 King rail, 128
 Kingfisher, belted, 69
 Kinglet, golden-crowned, 55
 Kingsnake, scarlet, 127
Kirchneriella lunaris, 34, 35
K. obesa, 34, 35
 Kirtland's snake, 127
 Kite
 Mississippi, 127
 swallow-tailed, 130
Koeleria macrantha, 120
 KOGER, MATTHEW E., 169
Kricogonia lyside, 87
Krigia occidentalis, 120
 KUMAR, K. S., 169
- Lace-winged Roadside Skipper, 86
 LACKI, MICHAEL J., 50
 Ladies'-tresses
 Great Plains, 122
 shining, 122
 sweetscent, 122
 yellow nodding, 122
 Lady's-slipper
 Kentucky, 118
 showy, 119
 small white, 118
 small yellow, 119
 Lady
 American, 87
 painted, 87
Laetilia fiskeella, 107
 LAINE, SEPPO, 170
 Lake chubsucker, 126
 Lake cress, 117
- Lake sturgeon, 125
Lampetra appendix, 126
 Lamprey
 chestnut, 126
 mountain brook, 126
 northern brook, 126
 southern brook, 126
Lampropeltis triangulum elapsoides, 127
Lampsilis abrupta, 124
L. ovata, 124
 Large bur-reed, 122
 Large orange sulphur, 87
 Large sedge, 118
 Large spotted St. John's-wort, 120
 Largeleaf grass-of-parnassus, 121
 Lark sparrow, 127
 Larkspur, Carolina, 119
Lasmigona compressa, 124
L. subviridis, 124
Lathyrus palustris, 120
L. venosus, 120
 LAU, JOANN M., 168
 Layside sulphur, 87
 Lead, effects of on rats, 108–114
 Leafcup, Tennessee, 121
 Leafshell, 130
 Cumberland, 130
 Leafwing, goatweed, 87
 Least bittern, 127
 Least darter, 130
 Least flycatcher, 127
 Least madtom, 126
 Least shrew, 164
 Least skipper, 86
 Least tern, 128
 Least trillium, 123
 Ozark, 123
 Least weasel, 128
 Leather-flower, blue jasmine, 118
Leavenworthia exigua var. *laciniata*, 120
L. torulosa, 120
Leiophyllum buxifolium, 120
 Lemming, southern bog, 163, 164
Lenthus sp., 21
 Leonard's skipper, 86
 Leopard frog, northern, 127
 Lepidoptera, 105–107
Lepidostoma sp., 21
 Lepidostomatidae, 21
Lepomis macrochirus, 75
L. macrochirus, 100
L. marginatus, 126
L. miniatus, 126
Lepotes marina, 87
Leptochloa fascicularis, 167
Leptodea leptodon, 130
Leptophlebiid mayfly, 125
 Leptophlebiidae, 20
Leptoxis praerosa, 123
Lerema accius, 86
Lerodea eufala, 86
 Lescur's bladderpod, 120

- Lespedeza capitata*, 120
L. stuvei, 120
Lesquerella globosa, 120
L. lescurii, 120
 Lesquereux's bladderpod, 120
 Lesser Adams cave beetle, 125
 Lettuce-leaf saxifrage, 122
Leucania calidior, 107
Leucothoe recurva, 120
Leucrocota, 15
L. prob. thetis, 20
Leuctra, 10, 13, 15
L. sp., 20
Leuctridae, 20
Lexingtonia dolabelloides, 124
Liatris cylindracea, 120
Libythaena carinenta bachmanii, 87
 Lichens, 117
Lilium philadelphicum, 120
L. superbum, 120
 Lilliput
 purple, 124
 Texas, 124
 Lily
 calla, 27
 Turk's cap, 120
 wood, 120
 Lily-of-the-valley
 American, 118
 wild, 120
Limenitis archippus, 87
L. arthemis arthemis, 87
L. arthemis astyanax, 87
 Limestone cave beetle, 125
 Limnephilid caddisfly, 125
Limnephilidae, 21
Limnobium spongia, 120
Limnophila sp., 22
Limnophyes sp., 22
 Limestone fameflower, 122
 Lip fern
 Alabama, 118
 Fée's, 118
Liparis loeselii, 120
Liriodendron tulipifera, 11, 54
Listera australis, 120
L. smallii, 120
Lithasia armigera, 123
L. geniculata, 123
L. salebrosa, 123
L. verrucosa, 123
Litobrantha recurvata, 125
 Little blue heron, 127
 Little glassywing, 86
 Little spectaclecase, 124
 Little wood satyr, 87
 Little yellow, 87
 Littlewing pearlymussel, 124
 Lizard, eastern slender glass, 127
Lobelia appendiculata var. *gattingeri*, 120
L. flabellaris, 27
L. nuttallii, 120
Lobelia
 Gattinger's, 120
 Nuttall's, 120
 Locust
 black, 54, 56
 water, 119
 Loesel's twayblade, 120
 Logperch, 70
 blotchside, 130
Lolium perenne, 167, 168
 Long-bract green orchis, 118
 Long-eared owl, 127
 Long-styled rush, 120
 Long-tailed shrew, 128
 Long-tailed skipper, 86
 Longhead darter, 126
 Longhorn beetle, sixbanded, 125
 Longleaf stitchwort, 122
 Longnose dace, 126
 Longsolid, 124
Lonicera dioica var. *orientalis*, 120
L. reticulata, 120
 Loosestrife
 Fraser's, 120
 trailing, 120
Lophodytes cucullatus, 127
Lordithon niger, 125
Lordithon rove beetle, black, 125
Lota lota, 126
 Louisiana broomrape, 121
 Louisville cave beetle, 125
 Louisville crayfish, 124
 Louisewort, swamp, 121
 Low rough aster, 117
 Low-tech teaching methods, 170
 open mindedness to, 170
 Lucy Braun's white snakeroot, 117
Ludwigia hirtella, 120
Ludwigia hairy, 120
Lumbricidae, 1–5
Lumbriculidae, 20
Lumbricus, 3
Luperina trigona, 107
Lycaena hyllus, 87
Lycaena phlaeas americana, 87
Lycopodiella appressa, 120
L. inundata, 120
Lycopodium clavatum, 120
Lygropia tripunctata, 106
Lype diversa, 21
Lysimachia fraseri, 120
L. radicans, 120
L. terrestris, 120
Lyctosis permagnaria, 125

Macrhybopsis gelida, 126
M. meeki, 126
Macrobrachium ohione, 124
M. rosenbergii, 165–167
Macroclmys temminckii, 127
 Macroinvertebrate communities,
 10–22
 Madtom
 brown, 126
 least, 126
 northern, 126
 slender, 126
Magnolia acuminata, 54
Maianthemum canadense, 120
M. stellatum, 120
 Maidenhair fern, southern, 117
Malirekus hastatus, 20
 Mallow, hispid false, 120
 MALPHRUS, BENJAMIN K., 133
Malus angustifolia, 120
Malvastrum hispidum, 120
 Mammals, 128, 130
 Mammoth Cave shrimp, 124
 Mandarin, nodding, 119
 Manna grass
 pale, 123
 sharp-scaled, 119
Manophylax butleri, 125
 Maple
 red, 54, 56
 sugar, 54
 Mapleleaf, winged, 130
 Marble, Olympia, 87
 Marigold, marsh, 130
 Marine blue, 87
 MARKER, GLENDA, 168
 MARKEY, M., 170
 Marsh marigold, 130
 Marsh-pink, slender, 122
Marshallia grandiflora, 120
 Maryland glyph, 123
 Masked shrew, 128
Masticophis flagellum flagellum, 130
Matelea carolinensis, 120
 Mathematics, 170
 Matricary grapefern, 117
 Matted feather moss, 117
 Mayfly
 burrowing, 125
 ephemerellid, 125
 heptageniid, 125
 leptophlebiid, 125
 robust pentagenian burrowing,
 130
 McCALL, MAUREEN, 168
 McDONOUGH, E., 170
 Meadow fritillary, 87
 Meadow vole, 164
 Meadow-parsnip, cutleaf, 123
 Meadowsweet, narrow-leaved, 122
Megaceryle alcyon, 71
 Megalitter, 171
 an Appalachian deformity, 171
Megisto cymela, 87
Melampyrum lineare var. *latifolium*,
 120
M. lineare var. *pectinatum*, 120
Melanostoma, 155
Melanthera nivea, 120
Melanthium parviflorum, 120
M. virginicum, 120
M. woodii, 120
Meleagris gallopavo, 53
Melithreptus, 155

- Menidia beryllina*, 126
 Mercury spurge, 119
 Merganser, hooded, 127
 Meropleon cosmion, 107
 Mesomphix rugeli, 123
 Metalmark
 northern, 87
 swamp, 87, 125
 Mice, radioprotective drug combination in, 167–170
 Michaux's bluets, 119
 Michaux's saxifrage, 122
 Micrasterias americana, 40, 43
 M. apiculata, 43, 44
 M. denticulata, 43, 44
 M. laticeps, 43, 44
 M. pinnatifida, 43, 44
 M. truncata, 44
 Micropsectra sp., 22
 Microspora pachyderma, 36, 38
 Microsporales, 36
 Microtendipes pedellus gp., 22
 M. rydalensis gp., 22
 Microtus ochrogaster, 163, 164
 M. pennsylvanicus, 164
 M. pinetorum, 162, 164
 Midland smooth softshell, 127
 Milbert's tortoise shell, 87
 Milkwort
 cross-leaf, 121
 Nuttall's, 121
 racemed, 121
 Mimulus floribundus, 96
 Minnow
 cypress, 126
 plains, 126
 stargazing, 126
 Minuartia cumberlandensis, 120
 M. glabra, 120
 Mirabilis albida, 120
 Mississippi green water snake, 127
 Mississippi kite, 127
 Missouri arrow-wood, 123
 Missouri rock-cress, 117
 Mniotilta vara, 53, 56
 MOBLEY, JOEL, 169
 Mock bishop's-weed, 121
 eastern, 121
 Nuttall's, 121
 Mock orange, 121
 hoary, 121
 Molanna, 15, 18
 M. blenda, 21
 Molannidae, 21
 Molothrus ater, 50, 54
 Monarch, 87
 Monarda punctata, 120
 Monkshood, blue, 117
 Monotropis odorata, 120
 Mountain birds, long-term conservation, 50–59
 Moorhen, common, 127
 MOORMAN, KENNETH M., 169
 Morehead radio telescope, 133–145
 first observations, 133–145
 MORGAN, ANN M., 166
 Morone
 chrysops \times M. saxatilis, 166
 Morus rubra, 54
 MORZILLO, ANITA T., 164
 Mosses, 117
 Moth
 geometrid, 125
 rattlesnake-master borer, 125
 Moths, Kentucky, 105–107
 Mottled duskywing, 86
 Mougeotia boodlei, 36, 38
 M. sphaerocarpa, 36, 38
 Mountain brook lamprey, 126
 Mountain creekshell, 124
 Mountain maple, 117
 Mountain woodsia, 123
 Mountain-lover, Canby's, 121
 Mountain-mint
 blunt, 121
 white leaved, 121
 Mourning cloak, 87
 Mourning dove, 53
 Mouse
 cotton, 128
 harvest, 163, 164
 house, 164, 171
 Moxostoma lacerum, 130
 M. poecilurum, 126
 M. valenciennesi, 130
 Mucket, pink, 124
 Mud snake, western, 127
 Mud-plantain
 blue, 119
 grassleaf, 119
 Muddy rocksnail, 123
 Mudminnow, central, 126
 Muhlenbergia bushii, 120
 M. cuspidata, 120
 M. glabriflora, 120
 Muhly
 Bush's, 120
 plains, 120
 MURPHY, D. SHANNON, 133
 Mus musculus, 164, 171
 Muscidae, 155
 Mussel
 oyster, 124
 salamander, 124
 Mussels, freshwater, 124, 130
 Mustela nivalis, 128
 Mycteria americana, 116
 Myotis austroriparius, 128
 M. grisescens, 128
 M. leibii, 128
 M. sodalis, 128
 Myotis
 eastern small-footed, 128
 gray, 128
 Indiana, 128
 southeastern, 128
 Myriophyllum heterophyllum, 121
 M. pinnatum, 121
 N-arginine dibasic convertase, 171
 stability of acidic domain of, 171
 Naiad, thread-like, 121
 Naididae, 20
 Najas gracillima, 121
 Nannyberry, 30–33
 Narrow-leaved bluecurls, 123
 Narrow-leaved meadowsweet, 122
 Nastra ilherminier, 86
 Nathalis iole, 87
 Neargyractis slossonalis, 106
 Neckera pennata, 117
 Necklace glade cress, 120
 Nemophila aphylla, 121
 Nemouridae, 20
 Neohelix dentifera, 123
 Neophylax, 15
 Neophylax sp., 21
 Nephopterix crassifasciella, 107
 N. vesustella, 106
 Nephrocystium obesum, 34, 44
 Nerodia cyclopion, 127
 N. erythrogaster neglecta, 127
 N. fasciata confluens, 127
 Nest density, artificial, 46–49
 effect on Canada Goose, 46–49
 Nestronia umbellula, 121
 Net-spinning caddisfly, Helma's, 125
 Netrium digitus, 36, 39
 Nettle-leaf noseburn, 123
 Nettle-leaf sage, 122
 New available names, moths, 105–107
 New York ironweed, 123
 NEWCOMB, ANGELA L., 169
 Nicrophorus americanus, 125
 Night-heron
 black-crowned, 127
 yellow-crowned, 127
 Nigronia, 14
 N. fasciatus, 21
 Nilotanyus sp., 22
 Nitrobacter, 27
 Nitrosomonas, 27
 Nocomis biguttatus, 126
 Noctuidae, 105, 107
 Nodding ladies'-tresses, yellow, 122
 Nodding mandarin, 119
 Nodding rattlesnake-root, 121
 Northern bog club-moss, 120
 Northern broken-dash, 86
 Northern brook lamprey, 126
 Northern cardinal, 53, 56
 Northern cavefish, 125
 Northern cloudywing, 86
 Northern coal skink, 127
 Northern crawfish frog, 127
 Northern dropseed, 122
 Northern fox grape, 123
 Northern hairstreak, 87, 125
 Northern harrier, 127
 Northern leopard frog, 127
 Northern madtom, 126
 Northern metalmark, 87

- Northern pearly-eye, 87
 Northern pike, 75
 Northern pine snake, 127
 Northern riffleshell, 124
 Northern shoveler, 127
 Northern starflower, 123
 Northern starhead topminnow, 126
 Northern white-cedar, 123
 Northern witch, 119
 Noseburn, nettle-leaf, 123
 NOTES, 163–164
 Notropis sp., 126
 N. albizonatus, 126
 N. hudsonius, 126
 N. maculatus, 126
 Noturus exilis, 126
 N. hildebrandi, 126
 N. phaeus, 126
 N. stigmosus, 126
 Nut-rush, fringed, 122
 Nuthatch, red-breasted, 55, 128
 Nuttall's lobelia, 120
 Nuttall's milkwort, 121
 Nuttall's mock bishop's-weed, 121
 Nyctanassa violacea, 127
 Nycticeius humeralis, 128
 Nycticorax nycticorax, 127
 Nyctiphyllax sp., 21
 Nymphalis antiopa, 87
 Nymphalis vaualbum j-album, 87
 Nyssa sylvatica, 54

 Obovaria retusa, 124
 Ocola skipper, 86
 Odonata, 21
 Oecophoridae, 105
 Oedogoniales, 36
 Oedogonium boscii, 36, 38
 O. capilliforme, 36, 38
 O. cardiacum, 36, 38
 O. grande, 36, 38
 Oenothera linifolia, 121
 O. oakesiana, 121
 O. perennis, 121
 O. triloba, 121
 Ohio shrimp, 124
 Oidaematophorus eupatorii, 107
 Old Well Cave beetle, 125
 Oldenlandia uniflora, 121
 Olethreutes tiliana, 106
 Oligia mactata, 107
 Oligochaeta, 1–5, 20
 Olivaceous sedge, 119
 Olive darter, 126
 Olympia marble, 87
 Oncophorus raii, 117
 One-flower fiddleleaf, 119
 Onosmodium molle ssp. hispidissimum, 121
 O. molle ssp. molle, 121
 O. molle ssp. occidentale, 121
 Onychonema laeve, 44
 Onyx rocksnail, 123
 Oocystis parva, 34, 35

 O. solitaria, 34, 35
 Ophiogomphus aspersus, 125
 O. howei, 125
 Ophisaurus attenuatus longicaudus, 127
 Ophorornis formosus, 53
 Orange sulphur, 87
 Orange tip, falcate, 87
 Orange, hoary mock, 121
 Orange, mock, 121
 Orange, sleepy, 87
 Orange-barred sulphur, 87
 Orangefood pimpleback, 124
 Orbesilum stipulatum, 130
 Orchard oriole, 53
 Orchid
 small purple-fingered, 121
 white fringeless, 121
 yellow-crested, 121
 Orchis, long-bract green, 118
 Orconectes burri, 124
 O. inermis, 124
 O. jeffersoni, 124
 O. lancifer, 124
 O. palmeri, 124
 O. pellucidus, 124
 Oriole, orchard, 53
 Ornate rocksnail, 123
 Orobanche ludoviciana, 121
 Oronectes australis, 124
 O. bisectus, 124
 Orontium aquaticum, 121
 Orthocladus annectens, 22
 Orthocyclops modestus, 165–166
 Orthotrichum diaphanum, 117
 Osprey, 128
 Ostrocerca prob. truncata, 20
 Oulimnius latiusculus, 21
 Ovate catchfly, 122
 Ovate fiddleleaf, 119
 Ovenbird, 53
 Owl
 barn, 128
 feeding habits, 163–164
 Owl
 long-eared, 127
 short-eared, 127
 Oxalis priceae, 121
 Oxydendrum arboreum, 54
 Oyster mussel, 124
 Ozark least trillium, 123

 Paintbrush, scarlet Indian, 118
 Painted lady, 87
 Painted trillium, 123
 Painted turtle, southern, 127
 Palaemonias ganteri, 124
 Palamedes swallowtail, 87
 PALAZZOLO, D. L., 169
 Pale false foxglove, 117
 Pale manna grass, 123
 Pale umbrella-wort, 120
 Palezone shiner, 126
 Pallid shiner, 126

 Pallid sturgeon, 126
 PALOMBI, PEGGY SHADDUCK, 169
 Palpomyia sp., 21
 Pale corydalis, 118
 Pandion haliaetus, 128
 PANEMANGALORE, MYNA, 108
 Panoquina ocola, 86
 Papaipema eryngii, 125
 Papershell, Cumberland, 124
 Papilio cresphontes, 87
 P. joanae, 87
 P. palamedes, 87
 P. polyxenes asterius, 87
 P. troilus, 87
 Papilo glaucus, 87
 Parachaetocladius sp., 22
 Paragnetina sp., 20
 Parakeet, Carolina, 130
 Paraleptophlebia, 10, 13, 15
 P. prob. ontario, 20
 Parametriocnemus, 15
 P. lundbecki, 22
 Parapediasia decorella, 106
 Parnassia asarifolia, 121
 P. grandifolia, 121
 Paronychia argyrocoma, 121
 Parrhasius m-album, 87
 Parsley, prairie, 130
 Parus bicolor, 53
 Parus carolinensis, 53
 Paspalum, bull, 121
 Paspalum boscianum, 121
 Passenger pigeon, 130
 Passerculus sandwichensis, 128
 Passerina cyanea, 53, 56
 Pastinaca sativa, 159
 Patera panselenus, 123
 Pawpaw, 165, 166
 controlled crosses, 165
 Kentucky State University project, 166
 molecular markers, 165
 Paxistima canbyi, 121
 Peachleaf willow, 122
 Peacock, white, 87
 Pearl crescent, 87
 Pearly-eye
 Creole, 87
 northern, 87
 southern, 87
 Pearlymussel
 cracking, 130
 dromedary, 130
 littlewing, 124
 slabside, 124
 Peatmoss, 117
 Peavine
 smooth veiny, 120
 vetchling, 120
 Peck's skipper, 86
 Pedastrum boryanum, 36, 37
 P. duplex, 36, 37
 P. simplex, 36, 37

- Pedicularis lanceolata*, 121
Pegias fabula, 124
Peltoperla arcuata, 20
Peltoperlidae, 20
Penium margaritaceum, 36, 39
Pennant, double-ringed, 125
Pennyroyal, rough, 119
Pentagenia robusta, 130
Pentagenian burrowing mayfly, robust, 130
Pepper and salt skipper, 86
Percina burtoni, 130
P. caprodes, 70
P. copelandi, 70
P. macrocephala, 126
P. squamata, 126
Percopsis omiscomaycus, 126
Peregrine falcon, 127
Perideridia americana, 121
Perlidae, 20
Perlodidae, 20
Peromyscus gossypinus, 128
Pesticide residue, 165
 in soil, 165
 measurement, 165
 mitigation, 165
 runoff, 165
Phacelia ranunculacea, 121
Phaeophyscia leana, 117
Phalacrocorax auritus, 128
Phalaris arundinacea, 23, 26, 27
Phenacobius uranops, 126
Pheucticus ludovicianus, 50
P. ludovicianus, 53, 128
Philadelphus inodorus, 121
P. pubescens, 121
PHILLIPS, B., 170
Philopotamidae, 21
Philtraea monillata, 107
Phlox bifida ssp. *bifida*, 121
P. bifida ssp. *stellaria*, 121
Phlox
 cleft, 121
 starry cleft, 121
Phoebe, eastern, 53
Phoebis agarithe, 87
P. philea, 87
P. sennae, 87
Pholisora catullus, 86
Photoacoustic measurements, 169
 in biological fluids, 169
 in biological tissues, 169
Phoxinus cumberlandensis, 126
Phyciodes batesii, 87, 125
P. tharos, 87
Physics & Astronomy, 170
Physiology & Biochemistry, 170–172
Physostegia intermedia, 130
Picea rubens, 52
Pickerel weed, 23, 26, 27
Pickerel, chain, 126
Pickerel-weed, 121
Picoides borealis, 128
P. pubescens, 53
P. villosus, 53
Pied-billed grebe, 128
Pieris rapae, 87
P. virginiensis, 87
Pigeon, passenger, 130
Pigmy rattlesnake, western, 127
Pigtoe
 pyramid, 124
 rough, 124
Pike, northern, 75
Pilaria sp., 22
Pilsbryna sp., 123
Pimpleback, orangefoot, 124
Pine Mountain tigersnail, 123
Pine snake, northern, 127
Pine, Virginia, 55
Pines, 52
Pinesap, sweet, 120
Pink mucket, 124
Pink, ring, 124
Pinus virginiana, 52, 54, 55
Pipevine swallowtail, 86
Pipilo erythrophthalmus, 53, 54
Piranga olivacea, 53
Pisidium sp., 20
Pituophis melanoleucus melanoleucus, 127
Plains frostweed, 119
Plains minnow, 126
Plains muhly, 120
Planariidae, 20
Planktosphaeria gelatinosa, 34, 35
Plant oils, effectiveness on air breathing insects, 166–171
Plantago cordata, 121
P. lanceolata, 167, 168
Plantain, heartleaf, 121
Plants, 117, 130
Platanthera cristata, 121
P. integrilabia, 121
P. psychodes, 121
Platygobio gracilis, 126
Plecoptera, 20
Plethadon hoffmani, 8
P. wehrlei, 8
Plethobascus cicatricosus, 130
P. cooperianus, 124
P. cyphus, 124
Plethodon cinereus, 6–9, 127
P. electromorphus, 6
P. richmondi, 6–9
P. shenandoah, 8
P. wehrlei, 127
Pleurobema clava, 124
P. oviforme, 124
P. plenum, 124
P. rubrum, 124
Pleurocera alveare, 123
P. curta, 123
Pleuroceridae, 60–63
Pleurotaenium constrictum, 39, 40
P. ehrenbergii, 39, 40
P. nodosum, 39, 40
Plukenet's cyperus, 118
Poa saltuensis, 121
Poaceae, 88
Poanes hobomok, 86
P. viator, 86
P. yehl, 86
P. zabulon, 86
Pocketbook, 124
 fat, 124
Podilymbus podiceps, 128
Podostemum ceratophyllum, 121
Pogonia, rose, 121
Pogonia ophioglossoides, 121
Poison sumac, 123
Polioptila caerulea, 53
Polites origenes, 86
P. peckius, 86
P. themistocles, 86
Polycentropodidae, 21
Polycentropus sp., 21
Polydamas swallowtail, 87
Polygala cruciata, 121
P. nuttallii, 121
P. paucifolia, 121
P. polygama, 121
Polygonia comma, 87
P. faunus, 125
P. faunus smythi, 87
P. intearrogationis, 87
P. progne, 87, 125
Polymnia laevigata, 121
Polypedilum aviceps gp., 22
P. convictum gp., 22
P. fallax gp., 22
P. haltare gp., 22
Polytaenia nuttalli, 130
Polytrichum pallidisetum, 117
P. piliferum, 117
P. strictum, 117
Pompeius verna, 86
POMPER, KIRK W., 165, 166
POND, GREGORY J., 10
Pondweed
 Illinois, 121
 spotted, 121
Pontederia cordata, 23, 26, 27, 121
Pontia protodice, 87
Poecetes gramineus, 128
Poppy-mallow, clustered, 118
Porcupine sedge, 118
Porter's reed, 118
Possum haw viburnum, 123
Potamilus capax, 124
Potamilus purpuratus, 124
Potamogeton illinoensis, 121
P. pulcher, 121
Potato-bean, Price's, 117
Potentilla erecta, 158
Potthastia sp., 22
Poultry by-product meal, 166
 in diets for fish, 166
Prairie gentian, 119
Prairie parsley, 130
Prairie redroot, 118
Prairie vole, 161, 164

- Prairie-chicken, greater, 130
 Prairie-clover, purple, 119
 Prawns, freshwater, 165, 166
 copepods as live food for, 165–166
 effect of water temperature on survival, 167
 transport density on survival, 165
 Pre-emphasis filter, auditory system model, 169
 Prenanthes alba, 121
 P. aspera, 121
 P. barbata, 121
 P. crepidinea, 121
 Pretty St. John's-wort, 120
 Price's potato-bean, 117
 Price's yellow wood sorrel, 121
 Prickly bog sedge, 118
 Primrose
 evening, 121
 yellow evening, 118
 Pristina aequisita, 20
 Privet, upland, 119
 Procambarus viaeviridis, 125
 Prosimulium sp., 22
 Prosopis, 155
 Prostate cancer cell lines, 170–171
 Proterometra albacauda, 103
 P. autraini, 102
 P. catenaria, 103
 P. dickermani, 102
 P. edneyi, 60–63, 103
 P. macrostoma, 99–104
 development in host, 99–104
 distome emergence, 99–104
 P. sagittaria, 103
 P. septimae, 103
 Prunus serotina, 54
 Psephenidae, 21
 Psephenus herricki, 21
 Pseudanophthalmus audax, 125
 P. calcareus, 125
 P. catoryctos, 125
 P. conditus, 125
 P. desertus major, 125
 P. frigidus, 125
 P. globiceps, 125
 P. horni abditus, 125
 P. horni caecus, 125
 P. horni horni, 125
 P. hypolithos, 125
 P. inexpectatus, 125
 P. parvus, 125
 P. pholeter, 125
 P. pubescens intrepidus, 125
 P. puteanus, 125
 P. rogersae, 125
 P. scholasticus, 125
 P. simulans, 125
 P. tenebrosus, 125
 P. troglodytes, 125
 Pseudolimnophila sp., 22
 Pseudoroegneria spicata, 92
 Pseudostenophylax uniformis, 21
 Psoralidium tenuiflorum, 121
 Pterophoridae, 105, 107
 Ptilimnium capillaceum, 121
 P. costatum, 121
 P. nuttallii, 121
 Ptychobranhus subtentum, 124
 Puma concolor cougar, 130
 Puma, eastern, 130
 Punctate coil, 123
 Purple giant hyssop, 117
 Purple lilliput, 124
 Purple prairie-clover, 119
 Purple sand grass, 123
 Purple, red spotted, 87
 Purple-fringed orchid, small, 121
 Purple-oat, 122
 Pussy willow, 122
 Pycnanthemum albescens, 121
 P. muticum, 121
 Pycnopsyche gentilis, 21
 Pycnopsyche prob. guttifer, 21
 Pygmy snaketail, 125
 Pyramid pigtoe, 124
 Pyrausta signatalis, 106
 Pyrgus centaureae, 86
 P. communis, 86
 P. wyandot, 125
 Pyrola americana, 121
 Quadrigula chodatii, 36, 37
 Quadrula cylindrica cylindrica, 124
 Q. fragosa, 130
 Q. tuberosa, 130
 Queen, 87
 Queen crater, 123
 Quercus alba, 11, 54
 Q. coccinea, 11
 Q. falcata, 54
 Q. macrocarpa, 89
 Q. mandanensis, 89
 Q. montana, 11
 Q. muehlenbergii, 54, 89
 Q. prinoides, 89
 Q. prinus, 54
 Q. rubra, 54
 Question mark, 87
 Quillwort
 blackfoot, 120
 Butler's, 120
 Rabbit-tobacco, small, 119
 Rabbitsfoot, 124
 Rabdotus dealbatus, 123
 Rabdotus, whitewashed, 123
 Racemed milkwort, 121
 Radio telescope, Morehead, 133–145
 Radioprotective drug combination in mice, 169–170
 Rafinesque's big-eared bat, 128
 Rail, king, 128
 Rainbow darter, 70
 Rallus elegans, 128
 Rana areolata circulosa, 127
 Rana pipiens, 127
 Rand's goldenrod, 122
 Ranunculaceae, 64
 Ranunculus ambigens, 121
 Raptioheptagenia cruentata, 125
 Rare biota of Kentucky, 115–132
 Rats
 adult, 108–114
 aged, 108–114
 body and tissue weights, 108–114
 effects of cadmium on, 108–114
 effects of lead on, 108–114
 effects of zinc on, 108–114
 weanling, 108–114
 Rattlesnake, western pigmy, 127
 Rattlesnake-master borer moth, 125
 Rattlesnake-root
 barbed, 121
 nodding, 121
 rough, 121
 white, 121
 Raven, common, 127
 Rayed bean, 124
 Red admiral, 87
 Red buckeye, 117
 Red elderberry, 122
 Red maple, 54, 56
 Red spotted purple, 87
 Red spruce, 52
 Red turtlehead, 118
 Red wolf, 130
 Red-backed vole, Kentucky, 128
 Red-banded, 87
 Red-breasted nuthatch, 55, 128
 Red-cockaded woodpecker, 128
 Red-eyed vireo, 54, 55
 Red-winged blackbird, 164
 Redback salamander, 127
 Redhorse
 blacktail, 126
 greater, 130
 Redroot, prairie, 118
 Redspotted sunfish, 126
 Redstart, American, 56
 Reed bent grass, 118
 Reed canary grass, 23, 26
 Reed grass, blue-joint, 118
 Porter's, 118
 REED, EDDIE B., 166
 REEDER, BRIAN C., 46
 Regal fritillary, 87, 125
 Regululus satrapa, 55
 Reithrodontomys megalotis, 164
 Reithrodontomys spp., 163
 Relict darter, 126
 Reniform sedge, 118
 Reptiles, 127, 130
 Rheotanytarsus sp., 22
 Rhinichthys cataractae, 126
 Rhodacme elatior, 123
 Rhododendron canescens, 121
 Rhyacionia aktita, 106
 Rhyacophila carolina, 21
 R. invaria gp., 21

- R. torva*, 21
R. vibox, 21
 Rhyacophilidae, 21
Rhynchosia tomentosa, 121
Rhynchospora globularis, 121
R. macrostachya, 121
Rhyngia, 155
 Ribbon snake
 eastern, 127
 western, 127
 RICE, DAVID, 169
 Rice
 Indian wild, 123
 southern wild, 123
 Riffleshell
 angled, 130
 northern, 124
 tan, 130
 Tennessee, 130
 Wabash, 130
 Rigid sedge, 118
 Ring pink, 124
Riparia riparia, 128
 River bulrush, 122
 Roan Mountain goldenrod, 122
 Roan sedge, 118
Robinia pseudoacacia, 54
 ROBINSON, DAVID L., 167, 168
 Robust pentagenian burrowing may-
 fly, 130
 Rock skullcap, 122
 Rock-cress
 Braun's, 117
 hairy, 117
 Missouri, 117
 Rockcastle aster, 117
 Rockshell, rough, 130
 Rocksnail
 armored, 123
 muddy, 123
 onyx, 123
 ornate, 123
 varicose, 123
 Roger's cave beetle, 125
 Rosaceae, 146–162
 Rose pogonia, 121
 Rose turtlehead, 118
 Rose verbena, 119
 Rose-breasted grosbeak, 50, 53, 55,
 56, 128
 Rosemary, Cumberland, 118
 ROSEN, RONALD, 62, 99
 Rosinweed, Appalachian, 122
 Rosinweed, tansy, 122
 Rosy twistedstalk, 122
 ROTH, LAUREN, 62
 Rough aster, low, 117
 Rough dropseed, 122
 Rough pennyroyal, 119
 Rough pigtoe, 124
 Rough rattlesnake-root, 121
 Rough rockshell, 130
 Round combshell, 130
 Round-head bush-clover, 120
 Round-headed cave beetle, 125
 Roundleaf fameflower, 123
 Rove beetle, black lordithon, 125
 Royal catchfly, 122
Rubus canadensis, 122
R. whartoniae, 122
 Ruby-throated hummingbird, 53
Rudbeckia subtomentosa, 122
 Rufous-sided towhee, 54, 56
 Rugged hornsnail, 123
 Running buffalo clover, 123
 Running-pine, 120
 Rush
 bog, 120
 jointed, 120
 long-styled, 120
Ruta graveolens, 159
 Rye, Svenson's wild, 119

Sabatia campanulata, 122
 Sachem, 86
 Sage, nettle-leaf, 122
Sagittaria graminea, 122
S. latifolia, 27
S. platyphylla, 122
S. rigida, 122
 Salamander mussel, 124
 redback, 127
 three-lined, 126
 Wehrle's, 127
 Salebriaria atratella, 106
S. tenebrosella, 106
Salix amygdaloides, 122
S. discolor, 122
S. nigra, 54
Salvia urticifolia, 122
Sambucus racemosa ssp. *pubens*,
 122
 Sand darter
 scaly, 130
 western, 125
 Sand grape, 123
 Sand grass, purple, 123
 Sand-myrtle, 120
 Sandpiper
 spotted, 127
 upland, 127
 Sandwort
 Appalachian, 120
 Cumberland, 120
Sanguisorba canadensis, 122
Santhidium armatum, 40, 42
 Sapsucker, yellow-bellied, 53
 Sassafras, 56
Sassafras albidum, 54, 56
 Satyr
 Carolina, 87
 gemmed, 87
 little wood, 87
Satyrium acadicum, 87
S. calanus falacer, 87
S. caryaevorum, 87
S. edwardsii, 87
S. favonius ontario, 87, 125

S. liparops, 87
S. titus mopsus, 87
Satyrodes appalachia, 87
 Savannah sparrow, 128
 Sawfin shiner, 126
Saxifraga michauxii, 122
S. micranthidifolia, 122
S. pensylvanica, 122
 Saxifrage
 brook, 118
 lettuce-leaf, 122
 Michaux's, 122
 swamp, 122
 Sayornis phoebe, 53
 Scaleshell, 130
 Scallopwing, Hayhurst's, 86
 Scaly sand darter, 130
Scaphirhynchus albus, 126
 Scarlet indian paintbrush, 118
 Scarlet kingsnake, 127
Scenedesmus bicaudatus, 36, 37
S. dimorphus, 36, 37
S. obtusus, 36, 37
S. quadricauda, 36, 37
Schisandra glabra, 122
Schizachne purpurascens, 122
 Scholarly cave beetle, 125
Schroederia setigera, 34, 35
 SCHUSTER, JESSICA, 62, 99
Schwalbea americana, 122
 Science Education, 171
 Scientists of Kentucky, 77–85
Scirpus expansus, 122
S. fluviatilis, 122
S. hallii, 122
S. heterochaetus, 122
S. microcarpus, 122
S. validus, 23, 26, 27
S. verecundus, 122
Scleria ciliata var. *ciliata*, 122
 Scorpion-weed, blue, 121
 Screwstem, yellow, 117
 Scrophulariaceae, 96
 Sculpted glyph, 123
 Scurf-pea stipuled, 130
 few-flowered, 121
Scutellaria arguta, 122
S. saxatilis, 122
 Sedge wren, 127
 Sedge
 Appalachian, 118
 bristly, 118
 broadwing, 118
 brown bog, 118
 cedar, 118
 Coastal Plain, 118
 Crawe's, 118
 cypress-swamp, 118
 epiphytic, 118
 finely-nerved, 118
 Fraser's, 118
 large, 118
 olivaceous, 119
 porcupine, 118

- prickly bog, 118
- reniform, 118
- rigid, 118
- roan, 118
- stalkgrain, 118
- straw, 118
- summer, 118
- Tarheel, 118
- umbel-like, 118
- weak stellate, 118
- woolly, 118
- Sedum telephoides*, 122
- SEED, T. M., 169
- Seiurus aurocapillus*, 53
- Selenastrum gracile*, 34, 35
- Selenisa sueroides*, 107
- Semotilus atromaculatus*, 11
- September elm, 123
- Sessile-fruit arrowhead, 122
- Setophaga ruticilla*, 53, 56
- Shad, Alabama, 125
- Shaggy cavesnail, 123
- Shaggy hedge-hyssop, 119
- Sharp-scaled manna grass, 119
- Sharp-shinned hawk, 127
- Shawnee darter, 126
- Sheepnose, 124
- Shell galaxies, hydrogen gas in, 170
- SHIBER, JOHN G., 170
- Shiner
 - blacktail, 125
 - bluntface, 125
 - palezone, 126
 - pallid, 126
 - sawfin, 126
 - spottail, 126
 - taillight, 126
- Shining ladies'-tresses, 122
- Shooting-star, French's, 119
- Short's goldenrod, 122
- Short's hedge-hyssop, 119
- Short-eared owl, 127
- Shortleaf skeleton 119
- Shortspire hornsnail, 123
- Shoveler, northern, 127
- Showy gentian, 119
- Showy lady's-slipper, 119
- Shrew, 163
 - least, 164
 - long-tailed, 128
 - masked, 128
- Shrimp
 - Mammoth Cave, 124
 - Ohio, 124
- Sicklefin chub, 126
- Sida hermaphrodita*, 122
- Side-oats grama, 118
- Silene ovata*, 122
- S. regia*, 122
- Silky aster, barrens, 117
- Silphium sunflower*, 119
- Silphium laciniatum* var. *laciniatum*, 122
- S. laciniatum* var. *robinsonii*, 122
- S. pinnatifidum*, 122
- S. wasiotense*, 122
- Silver-bordered fritillary, 87
- Silver-spotted skipper, 86
- Silverbell, common, 119
- Silvering, 121
- Silvery aster, silvery, 117
- Silvery blue, 87
- Silvery checkerspot, 87
- Simpsonia ambigua*, 124
- Simuliidae, 22
- Simulium* sp., 22
- Sistrurus miliarius streckeri*, 127
- Sitanion hystrix*, 88, 89, 92
- S. minus*, 88
- Sitta canadensis*, 55, 128
- S. carolinensis*, 53
- Sixbanded longhorn beetle, 125
- Skeleton grass
 - bearded, 119
 - shortleaf, 119
- Skink
 - northern coal, 127
 - southeastern five-lined, 127
 - southern coal, 127
- Skipper
 - Appalachian grizzled, 125
 - Bell's roadside, 86
 - broadwinged, 86
 - clouded, 86
 - cobweb, 86
 - common checkered, 86
 - common roadside, 86
 - crossline, 86
 - Delaware, 86
 - Dion, 86
 - Duke's, 86, 125
 - Dun, 86
 - dusted, 86
 - Eufala, 86
 - European, 86
 - fiery, 86
 - gold-banded, 86
 - grizzled, 86
 - hobomok, 86
 - Indian, 86
 - lace-winged roadside, 86
 - least, 86
 - Leonard's, 86
 - long-tailed, 86
 - Ocala, 86
 - Peck's, 86
 - pepper and salt, 86
 - silver-spotted, 86
 - swarthy, 86
 - tawny-edged, 86
 - Yehl, 86
 - Zabulon, 86
- Skullcap
 - hairy, 122
 - rock, 122
- Skunk, eastern spotted, 128
- Slabside pearlymussel, 124
- Sleepy duskywing, 86
- Sleepy orange, 87
- Slender blazingstar, 120
- Slender bulrush, 122
- Slender dragon-head, 130
- Slender glass lizard, eastern, 127
- Slender madtom, 126
- Slender marsh-pink, 122
- Small enchanter's-nightshade, 118
- Small lady's-slipper, yellow, 119
- Small purple-fringed orchid, 121
- Small rabbit-tobacco, 119
- Small sundrops, 121
- Small white lady's-slipper, 118
- Small yellow lady's-slipper, 119
- Small-flower baby-blue-eyes, 121
- Small-flowered false hellebore, 120
- Small-flowered thoroughwort, 119
- Small-footed myotis, eastern, 128
- Small-fruit bulrush, 122
- Smallscale darter, 126
- SMITH, J., 171
- Smooth blackberry, 122
- Smooth softshell, midland, 127
- Smooth veiny peavine, 120
- Snail, 60–63, 121
- Snake
 - broad-banded water, 127
 - copperbelly water, 127
 - corn, 127
 - eastern ribbon, 127
 - Kirtland's, 127
 - Mississippi green water, 127
 - northern pine, 127
 - western mud, 127
 - western ribbon, 127
- Snakeroot
 - Lucy Braun's white, 117
 - white, 167
- Snaketail
 - brook, 125
 - pygmy, 125
- Snapping turtle, alligator, 127
- Snout-bean, hairy, 121
- Snow melanthera, 120
- Snow trillium, 123
- Snowberry, 122
- Snuffbox, 124
- Soft false gromwell, 121
- Soft-haired thermopsis, 123
- Softshell, midland smooth, 127
- Softstem bulrush, 23, 26, 27
- Solidago albopilosa*, 122
- S. buckleyi*, 122
- S. curtisii*, 122
- S. gracillima*, 122
- S. puberula*, 122
- S. roanensis*, 122
- S. shortii*, 122
- S. simplex* ssp. *randii*, 122
- S. squarrosa*, 122
- Solomon-seal, starry false, 120
- Sootywing, common, 86
- Sorastrum americanum*, 36, 37
- Sorex* spp., 163

- S. cinereus*, 128
S. dispar blitchi, 128
 Sorrel, Price's yellow wood, 121
 Southeastern five-lined skink, 127
 Southeastern myotis, 128
 Southern bog club-moss, 120
 Southern bog goldenrod, 122
 Southern bog lemming, 163
 Southern broken-dash, 86
 Southern brook lamprey, 126
 Southern cavefish, 126
 Southern cloudywing, 86
 Southern coal skink, 127
 Southern crabapple, 120
 Southern dogface, 87
 Southern heartleaf, 119
 Southern maidenhair fern, 117
 Southern painted turtle, 127
 Southern pearly-eye, 87
 Southern shield wood fern, 119
 Southern twayblade, 120
 Southern wild rice, 123
Soyedina sp., 20
Sparganium eurycarpum, 120
 Sparrow
 Bachman's, 127
 Henslow's, 127
 lark, 127
 Savannah, 128
 vesper, 128
 Spearwort, water-plantain, 121
 Speckled darter, 70
 Spectaclecase, 124
 little, 124
 Speedwell, American, 123
Speyeria aphrodite, 87
S. cybele, 87
S. diana, 87
S. idalia, 87, 125
Sphaeriidae, 20
Sphaerocystis schroeteri, 34, 35
Sphaerosoma aubertianum, 44
Sphagnum quinquefarium, 117
Sphenopholis pensylvanica, 122
Sphyrapicus varius, 53
 Spicebush swallowtail, 87
 Spiked hoary-pea, 123
Spilogale putorius, 128
 Spinulose wood fern, 119
Spiraea alba var. *alba*, 122
S. virginiana, 122
Spiraea, Virginia, 122
Spiranthes lucida, 122
S. magnicamporum, 122
S. ochroleuca, 122
S. odorata, 122
Spirogyra communis, 36, 38
S. pratensis, 36, 38
S. varians, 36, 38
Spirotaenia condensata, 36, 39
Spondylosium moniliforme, 44
 Spoon-leaved sundew, 119
Sporobolus clandestinus, 122
S. heterolepis, 122
 Spottail shiner, 126
 Spotted beebalm, 120
 Spotted coralroot, 118
 Spotted darter, 126
 Spotted joe-pye-weed, 119
 Spotted pondweed, 121
 Spotted sandpiper, 127
 Spotted skunk, eastern, 128
 Spreading false foxglove, 117
 Spring azure, 87
 Spruce, red, 52
 Spurge, mercury, 119
 Squarrose goldenrod, 122
 SRINIVASAN, V., 169
 St. John's-wort
 creeping, 119
 large spotted, 120
 pretty, 120
 St. Peter's-wort, 119
Stachys eplingii, 122
 Stalkgrain sedge, 118
Staphylus hayhurstii, 86
 Star tickseed, 118
 Starflower, northern, 123
 Stargazing minnow, 126
 Starhead topminnow, northern, 126
 Starry cleft phlox, 121
 Starry false solomon-seal, 120
 Starvine, bay, 122
 State records, moths, 105–107
Staurostrum alternans, 40, 42
S. arcticon, 40, 42
S. botryophilum, 40, 42
S. brasiliense, 40, 42
S. chaetoceros, 40, 42
S. crenulatum, 40, 42
S. curvatum, 40, 42
S. dickiei, 40, 42
S. hexacerum, 40, 42
S. leptocladum, 40, 42
S. limneticum, 40, 42
S. setigerum, 40, 42
 Steele's joe-pye-weed, 119
Stellaria fontinalis, 122
S. longifolia, 122
 Stellate sedge, weak, 118
 Stemless evening-primrose, 121
Stenacron interpunctatum, 20
Stenelmis sp., 21
S. crenata, 21
Stenonema bednariki, 125
S. meririvulanum, 20
S. meririvulanum, 15
S. vicarium, 20
Stenoptilodes brevipennis, 107
Sterna antillarum, 128
 Stevens Creek Cave beetle, 125
 Stipuled scurf-pea, 130
 Stitchwort
 longleaf, 122
 water, 122
 Stonecrop, Allegheny, 122
 Stonefly, 18
 Stoneroller, central, 75
 Straw sedge, 118
Streptopus roseus var. *perspectus*, 122
 Striped bass hybrid, 166
 Striped hairstreak, 87
 Striped whitelip, 124
Strymon melinus, 87
 Sturgeon chub, 126
 Sturgeon
 lake, 125
 pallid, 126
Stygobromus vitreus, 125
Stylogomphus, 14
S. albistylus, 21
Stylurus notatus, 125
 Sucker
 blackfin, 126
 hairlip, 130
 Sugar maple, 54
 Sugarspoon, 130
 Sulphur
 clouded, 87
 cloudless, 87
 dainty, 87
 large orange, 87
 Layside, 87
 orange, 87
 orange-barred, 87
 Sumac, poison, 123
 Summer sedge, 118
 Sundew
 dwarf, 119
 spoon-leaved, 119
 Sundrops
 small, 121
 thread-leaf, 121
 Sunfish
 dollar, 126
 redspotted, 126
 Sunflower
 Eggert's, 119
 silphium, 119
 Sunshine bass, 166
 diets for, 166
 Superficial coset curiosities, 170
 Supplejack, 117
 Surprising cave beetle, 125
 Svenson's wild rye, 119
 Swallow
 bank, 128
 polydamas, 87
 Swallow-tailed kite, 130
 Swallowtail
 black, 87
 giant, 87
 Joan's, 87
 palamedes, 87
 pipevine, 86
 spicebush, 87
 tiger, 87
 zebra, 87
 Swamp darter, 126
 Swamp lousewort, 121
 Swamp metalmark, 87

- Swamp metalmark, 125
 Swamp saxifrage, 122
 Swamp wedgescale, 122
 Swamp-candles, 120
 Swarthy skipper, 86
 Sweet birch, 54
 Sweet coneflower, 122
 Sweet flag, 23, 26, 27
 Sweet pinesap, 120
 Sweet-fern, 118
 Sweetscent ladies'-tresses, 122
 Sweetshrub, 118
 Sweltsa, 15
S. sp., 20
Symphoricarpos albus, 122
Synaptomys cooperi, 163, 164
Synclita tinealis, 106
Synorthocladus sp., 22
 Syritta, 155
 Syrphidae, 155
 Syrphus, 155
- Taillight shiner, 126
Talinum calcaricum, 122
 Tall beaked-rush, 121
 Tall bush-clover, 120
 Tall fescue, 27
 Tall hairy groovebur, 117
Talium teretifolium, 123
 Tamoxifen, 170–171
 effect on Cathepsin B, 170–171
 effect on Cathepsin L, 170–171
 effect on cysteine proteases, 170–171
 Tan riffleshell, 130
 Tansy rosinweed, 122
 Tanytarsus, 15
 Tanytarsus *sp.*, 22
Taraxacum officinale, 167, 168
 inheritance of morphological characteristics, 167
 inheritance of physiological characteristics, 167
 Tarheel sedge, 118
 Tatum Cave beetle, 125
 Tawny cotton-grass, 119
 Tawny crescent, 87, 125
 Tawny emperor, 87
 Tawny-edged skipper, 86
Taxus canadensis, 123
 Teal, blue-winged, 127
Teilingia excavata, 44
 Telescope, Morehead radio, 133–145
 Ten-lobed false foxglove, 117
 Tennessee aster, 117
 Tennessee clubshell, 124
 Tennessee leafcup, 121
 Tennessee riffleshell, 130
Tephrosia spicata, 123
 Tern
 black, 130
 least, 128
 Terrestrial vertebrate species, 171
 richness of, 171
Tetrasporales, 34
Tetmemorus brebissonii, 39, 40
Tetraedron minimum, 34, 35
T. regulare, 34, 35
 Texas aster, 117
 Texas lilliput, 124
Thalictrum clavatum, 62
T. mirabile, 62
 chromosome number of, 62–63
Thamnophis proximus proximus, 127
T. sauritus sauritus, 127
Thaspium pinnatifidum, 123
Theliopsyche, 15, 18
T. sp., 21
Thermopsis mollis, 123
 soft-haired, 123
Thienimanniella sp., 22
 THIERET, JOHN W., 146
Thoburnia atripinnis, 126
 THOGMARTIN, WAYNE E., 164
 THOMPSON, J., 170
 THOMPSON, KENNETH R., 166
 Thoroughwort, small-flowered, 119
Thorybes bathyllus, 86
T. confusus, 86
T. pylades, 86
 Thread-leaf sundrops, 121
 Thread-like naiad, 121
 Threadfoot, 121
 Three-awn grass, branched, 117
 Three-lined salamander, 126
 Three-toed amphiuma, 126
 Thrush, wood, 56
Thryomanes bewickii, 128
Thryothorus ludovicianus, 53, 56
Thuja occidentalis, 123
Thymelicus lineola, 86
 Tickseed, star, 118
 TIDWELL, JAMES H., 165, 166, 167
 Tiger swallowtail, 87
 Tigersnail, Pine Mountain, 123
Tilia spp., 54
Tillium nivale, 123
 Tippecanoe darter, 70
Tipula sp., 22
 Tipulidae, 22
 Topminnow
 golden, 126
 northern starhead, 126
Torreyochloa pallida, 123
 Tortoise shell, compton, 87
 Milbert's, 87
 Tortricidae, 105, 106
 Tortula, 117
Tortula norvegica, 117
 Towhee, rufous-sided, 54, 56
Toxicodendron vernix, 123
Toxolasma lividus, 124
T. texasiensis, 124
Tragia urticifolia, 123
 Trailing loosestrife, 120
Traverella lewisi, 125
 Treatment, wastewater, 23–29
 Treefrog
 barking, 127
 bird-voiced, 126
 gray, 127
 green, 127
 Trematode, digenetic, 60–63
Trepocarpus, 123
Trepocarpus aethusae, 123
 Trichoptera, 21
Trichostema setaceum, 123
 Tricladia, 20
Trientalis borealis, 123
Trifolium reflexum, 123
T. repens, 167, 168
T. stoloniferum, 92, 123
Trillium pusillum var. *ozarkanum*, 123
T. pusillum var. *pusillum*, 123
T. undulatum, 123
 Trillium
 least, 123
 Ozark least, 123
 painted, 123
 snow, 123
Triplasis purpurea, 123
Troglodytes troglodytes, 55
 Trout, American brook, 126
 Trout-perch, 126
Tryphlichthys subterraneus, 126
Tsuga canadensis, 11
 TUAN, VO-DINH, 169
 Tubercled blossom, 130
 Tubificidae, 20
 Tufted hair, 119
 Turbellaria, 20
 Turk's cap lily, 120
 Turkeybeard, eastern, 123
 Turtle
 alligator snapping, 127
 southern painted, 127
 Turtlehead
 red, 118
 rose, 118
Tvetenia bavarica gp., 22
Tvetinia discloripes gp., 22
 Twayblade
 kidney-leaf, 120
 Loesel's, 120
 southern, 120
 Twistedstalk, rosy, 122
Tympanuchus cupido, 130
Typha latifolia, 23–27
Tyto alba, 128, 163–164
- Uenoidae, 21
Ulmus serotina, 123
 Umbel-like sedge, 118
Umbra limi, 126
 Umbrella-wort, pale, 120
 Upland privet, 119
 Upland sandpiper, 127
 Urbanus proteus, 86

- Ursus americanus*, 128
Utricularia macrorhiza, 123

Vallisneria americana, 123
 VANARNUM, AARON, 165, 166, 167
Vanessa atalanta, 87
V. cardui, 87
V. virginensis, 87
 Variable-leaved heartleaf, 119
 Varicose rocksnail, 123
 Variegated fritillary, 87
 Vascular plants, 117–123
 Veery, 55
Veneroida, 20
Verbena, rose, 119
Vermivora bachmanii, 130
V. chrysoptera, 50, 53, 128
V. pinus, 55
Vernonia noveboracensis, 123
Veronica americana, 123
Vertigo
 cupped, 124
 delicate, 124
Vertigo bollesiana, 124
V. clappi, 124
Vesper sparrow, 128
Vetchling peavine, 120
Viburnum lentago, 30–33
V. molle, 123
V. nudum, 123
V. prunifolium, 30–32
V. rafinesquianum var. *rafinesquianum*, 123
V. rufidulum, 30–32
Viburnum, possum haw, 123
Viceroy, 87
Villosa fabalis, 124
V. lienosa, 124
V. ortmanni, 124
V. trabalis, 124
V. vanuxemensis, 124
Viola septemloba var. *egglestonii*, 123
V. walteri, 123
 Violet
 Eggleston's, 123
 Walter's, 123
Vireo bellii, 128
V. flavifrons, 53
V. griseus, 53
V. olivaceus, 53, 54
V. solitarius, 53, 54
Vireo
 Bell's, 128
 blue-headed, 55
 red-eyed, 54, 55
 white-eyed, 53
Virginia big-eared bat, 128
Virginia bladetooth, 123
Virginia bunchflower, 120
Virginia pine, 55
Virginia spiraea, 122
Virginia waterleaf, 119
Virginia-mallow, 122
 Vitamin E, 170–171
 effect on Cathepsin B, 170–171
 effect on Cathepsin L, 170–171
 effect on cysteine proteases, 170–171
 VITATOE, LEIGH ANNE, 167
Vitis labrusca, 123
V. rupestris, 123
Vitrinizonites latissimus, 124
 Volatile emissions
 biological effects of, 167–168
 from cut turf, 167–168
 Vole
 Kentucky red-backed, 128
 meadow, 164
 prairie, 163, 164
 woodland, 163, 164
 Volvocales, 34

 Wabash riffleshell, 130
 WALCK, JEFFREY L., 63
Wallengrenia egeremet, 86
W. otho, 86
 Walnut, white, 120
 Walter's violet, 123
 Warbler
 Bachman's, 130
 black-and-white, 56
 black-throated blue, 54
 black-throated blue, 55
 black-throated blue, 56
 Blackburnian, 50, 53, 55, 127
 blue-winged, 55
 Canada, 50, 53, 55, 128
 cerulean, 50, 53
 chestnut-sided, 54–56
 golden-winged, 50, 53, 55, 128
 hooded, 54–56
 yellow-rumped, 53
 WARNER, RICHARD C., 23
 Wartyback, white, 130
 Wastewater treatment, 23–29
 Water hickory, 118
 Water locust, 119
 Water snake
 broad-banded, 127
 copperbelly, 127
 Mississippi green, 127
 Water stitchwort, 122
 Water-milfoil
 broadleaf, 121
 cutleaf, 121
 Water-pennywort, American, 119
 Water-plantain spearwort, 121
 Water-purslane, 119
 Waterleaf, Virginia, 119
 Waterweed, 119
 Weak stellate sedge, 118
 Weasel, least, 128
Webbhelix multilineata, 124
 WEBSTER, CARL D., 166
 WECKMAN, TIMOTHY J., 30
 Wedge-leaf whitlow, 119
 Wedgescale, swamp, 122
 Wehrle's salamander, 127
 WEIBEL, CHARLES, 165, 166
 WESSEL, MARK V., 146
 West Virginia white, 87
 Western dwarf dandelion, 120
 Western false gromwell, 121
 Western mud snake, 127
 Western pigmy rattlesnake, 127
 Western ribbon snake, 127
 Western sand darter, 125
 Westland size, effect on Canada Goose, 46–49
 Wetlands, constructed, 23–29
 Wharton's dewberry, 122
 White admiral, 87
 White catpaw, 130
 White fringeless orchid, 121
 White heath aster, 117
 White laldy's slipper, small, 118
 White peacock, 87
 White rattlesnake-root, 121
 White snakeroot, 167
 White snakeroot, Lucy Braun's, 117
 White walnut, 120
 White wartyback, 130
 White
 cabbage, 87
 checkered, 87
 West Virginia, 87
 White-cedar, northern, 123
 White-eyed vireo, 53
 White-haired goldenrod, 122
 White-leaved mountain-mint, 121
 White-m, 87
 Whitelip, big-tooth, 123
 Whitelip, striped, 124
 WHITEMAN, HOWARD, 171
 Whitewashed rabdotus, 123
 Whitlow- wedge-leaf, 119
 Whorled aster, 119
 Whorled horse-balm, 118
 Wild honeysuckle, 120
 Wild Indigo duskywing, 86
 Wild indigo
 blue, 117
 cream, 117
 yellow, 117
 Wild lily-of-the-valley, 120
 Wild rice
 Indian, 123
 southern, 123
 Wild rye, Svenson's, 119
 Willow, 55
 peachleaf, 122
 pussy, 122
Wilsonia canadensis, 50, 53, 128
W. citrina, 53
 Winged mapleleaf, 130
 Winter wren, 55
 Wintergreen, American, 121
 Wire fern moss, 117
 Witch northern, 119
 Wolf

- gray, 130
 red, 130
 Wood fern
 southern shield, 119
 spinulose, 119
 Wood lily, 120
 Wood sorrel, Price's yellow, 121
 Wood thrush, 56
 Wood-nymph, common, 87
 Woodland beak-rush, 122
 Woodland vole, 163, 164
 Woodpecker
 ivory-billed, 130
 red-cockaded, 128
 Woodsia appalachiana, 123
 Woodsia, mountain, 123
 Woolly sedge, 118
 Wormaldia prob. moesta, 21
 Wren
 Bewick's, 128
 Carolina, 56
 sedge, 127
 winter, 55
 WRIGHT, DONALD J., 105
 Wrinkled button, 123

 Xanthidium antilopaeum, 40, 42
 Xerophyllum asphodeloides, 123
 Xyris difformis, 123

 Yandell, David Wendel, M. D., 77–85
 Yehl skipper, 86
 Yellow blossom, 130
 Yellow evening primrose, 118
 Yellow gentian, 119
 Yellow nodding ladies'-tresses, 122
 Yellow screwstem, 117
 Yellow water l., 27
 Yellow wild indigo, 117
 Yellow wood sorrel, Price's, 121
 Yellow, little, 87
 Yellow-bellied sapsucker, 53
 Yellow-billed cuckoo, 53

 Yellow-breasted chat, 53
 Yellow-crested orchid, 121
 Yellow-crowned night-heron, 127
 Yellow-eye, Carolina, 123
 Yellow-poplar, 54, 56
 Yellow-rumped warbler, 53
 Yellowthroat, common, 53
 Yew, Canadian, 123
 Yugus, 18
 Yugus sp., 20

 Zabulon skipper, 86
 Zantedeschia aethiopica, 27
 Zarucco duskywing, 86
 Zebra swallowtail, 87
 Zenaida macroura, 53
 Zenaida macroura, 53
 Zinc, effects of on rats, 108–114
 Zizania palustris var. interior, 123
 Zizaniopsis miliacea, 123
 Zoology & Entomology, 171
 Zygnema decussatum, 36, 38
 Zygnematales, 36



CONTENTS

ARTICLES

Conservation Status and Nesting Biology of the Endangered Duskytail Darter, <i>Etheostoma Percnorum</i> , in the Big South Fork of the Cumberland River, Kentucky. <i>David J. Eisenhour and Brooks M. Burr</i>	67
Scientists of Kentucky David Wendel Yandell, M. D. <i>Nancy Disher Baird</i>	77
A Field Checklist of Kentucky Butterflies. <i>Charles V. Covell Jr.</i>	86
Notes on North American <i>Elymus</i> Species (Poaceae) with Paired Spikelets: I. <i>E. macgregorii</i> sp. nov. and <i>E. glaucus</i> ssp. <i>mackenzii</i> comb. nov. <i>Julian J. N. Campbell</i>	88
<i>Proterometra macrostoma</i> (Digenea: Azygiidae): Distome Emergence From the Cercarial Tail and Subsequent Development in the Definitive Host. <i>Ronald Rosen, Kelly Adams, Emilia Boiadgieva, and Jessica Schuster</i>	99
New State Records and New Available Names for Species of Kentucky Moths (Insecta: Lepidoptera). <i>Charles V. Covell Jr., Loran D. Gibson, and Donald J. Wright</i>	105
Comparative Effects of Zinc, Lead, and Cadmium on Body and Tissue Weights of Weanling, Adult, and Aged Rats. <i>F. N. Bebe and Myna Panemangalore</i>	108
Rare and Extirpated Biota of Kentucky. <i>Kentucky State Nature Preserves Commission</i>	115
First Observations with the Morehead Radio Telescope, Morehead State University, Morehead, Kentucky. <i>Benjamin K. Malphrus, Michael S. Combs, Michael A. James, D. Shannon Murphy, D. Kevin Brown, Jeff Kruth, and R. Douglas Kelly</i>	133
<i>Agrimonia</i> (Rosaceae) in Kentucky with Notes on the Genus. <i>Mark V. Wessel and John W. Thieret</i>	146

NOTE

Barn Owl (<i>Tyto alba</i>) Feeding Habits at Yellowbank Wildlife Management Area, Breckinridge County, Kentucky. <i>Anita T. Morzillo, Hetti A. Brown, Wayne E. Thogmartin, and Jennifer H. Herner-Thogmartin</i>	163
Abstracts of Some Papers Presented at the 1999 Meeting of the Kentucky Academy of Science	165
List of Reviewers for Volume 61	173
Index to Volume 61	174